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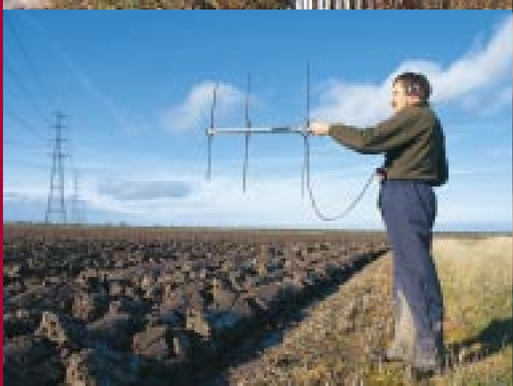


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The ecological impact of sweet chestnut coppice silviculture on former ancient, broadleaved woodland sites in south-east England

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Preface

Sweet chestnut *Castanea sativa* is treated as an "honorary native" in some woods and as an undesirable alien in others. A considered view of its impact on nature conservation values has been hampered by the lack of an up-to-date review of its ecology. English Nature is grateful to the authors and sponsors of this report for permission to reproduce it in our research report series.

Keith Kirby
English Nature, Peterborough

Acknowledgements

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Thanks must also go to the delegates who attended and contributed to the workshop held at Department of Agricultural Sciences campus of Imperial College in February 2003, particularly our invited speakers and workshop facilitators. We are grateful for contributions to our review report from Debbie Bartlett, David Rossney (ESUS Forestry & Woodlands Ltd), Ralph Harmer (Forestry Commission), Graham Bull (Forestry Commission), Keith Kirby (English Nature), John Badmin (Kent Field Club), David Gardner and Mark Parsons (Butterfly Conservation). We would also like to thank the Bat Conservation Trust for granting us permission to include four bat distribution maps, Nigel Braden and Karen Russell for their sweet chestnut distribution map and Pamela Howell for the title page and interleaf illustrations.

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Summary

Sweet chestnut *Castanea sativa* was probably introduced into Britain over two millennia ago, since when its range has expanded steadily across southern England as a result of increasing domestication and naturalization. Initially valued for its nuts and timber properties, planting of pure chestnut stands for coppice accelerated sharply in the nineteenth century in response to strong hop-growing and fencing markets. These plantations were often located on sites of semi-natural, broadleaved woodlands that were comprehensively cleared of competing woody species. Current woodland census data underestimates the area of chestnut in Britain at c.19,000 ha, while the true area of chestnut-dominated woodland could be twice this extent.

Chestnut is well adapted to a wide range of site and soil conditions normally occupied by mixed broadleaved woodland communities characterised by oak, hazel, birch and ash. Its ecological impact on these communities is reviewed according to the main taxonomic groups: vegetation, fungi, invertebrates, birds and mammals. An overall conclusion is that few, if any species recorded so far within these major groups are directly dependent on chestnut as a host. While chestnut does support a number of species that also occur on trees and shrubs comprising the equivalent native broadleaved community, the number and variety associated with the former appears to be lower, especially in monoculture stands.

On the other hand, the maintenance of the coppice cycle in commercially viable chestnut crops can be beneficial to some notable species that are dependent upon young growth stages, such as fritillary butterflies and migrant birds, while the system of relatively small coupe sizes and extensive ride networks present in worked coppice adds diversity at the whole forest scale. However, as the uncertain future of markets for chestnut shows, there is no guarantee of consistent or continued working, except perhaps on sites already in nature conservation ownership. Where management incentives are available to diversify chestnut sites, alternatives include varying the rotation length, reducing the dominance of the chestnut canopy by stool removal and/or thinning, reintroducing native species, promoting stands to high forest and practising minimum intervention. These techniques are most appropriate on sites where chestnut substitution is recent, rather than where historical accounts and evidence on the ground confirm a long and continuous presence of the species.

Apart from the effects of market uncertainty, the future status of chestnut will be determined by its resilience to new challenges from disease and climate change. The recent advance of chestnut blight *Cryphonectria parasitica* in northern Europe suggests strongly that it will extend its range into Britain; the impact could be to reduce the dominance of chestnut in woodlands in southern England. Conversely, scenarios predicted for climate warming during the remainder of the 21st century indicate that conditions favouring chestnut will be enhanced, causing it to become a more invasive and aggressive component of ancient woodland.

A number of recommendations are made for further research and study.

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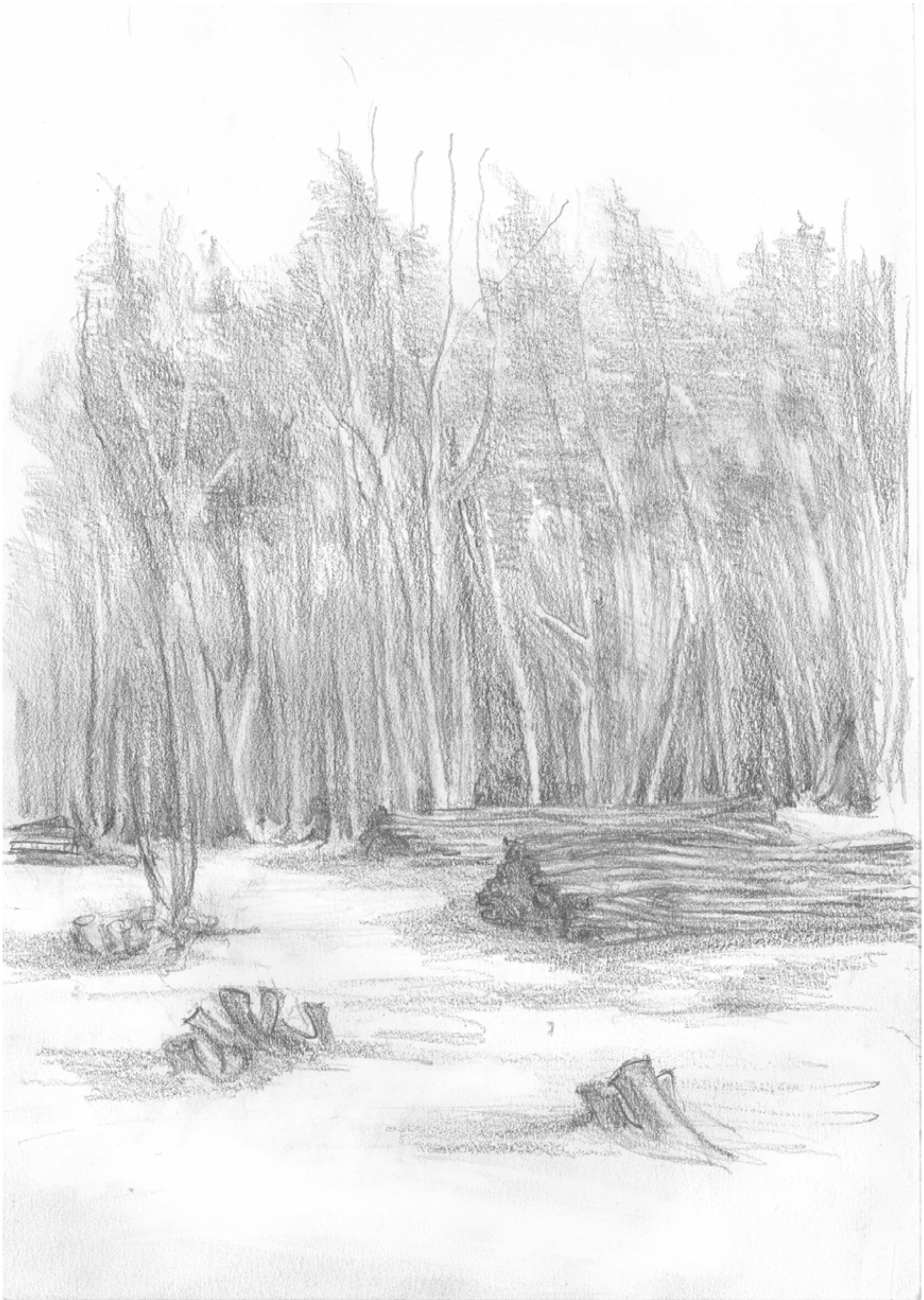

1. Introduction

1.1 Aim

Sweet chestnut is not native to the UK but is widely distributed in southern England, where it exerts dominance over significant areas of semi-natural, broadleaved woodland. In order to better understand the ecological impact of the species, the Department of Agricultural Sciences at Imperial College was invited by two sponsors, Kent County Council and the Gallagher Group respectively, to review the scientific literature relating to chestnut in Britain. Our remit was *to review available evidence for or against the hypothesis that the presence of sweet chestnut coppice on sites of former, semi-natural broadleaved woodland, results in an impoverishment in species diversity and a decline in nature conservation interest.* Our objectives in carrying out the review were to:

- characterise the site types and woodland communities in which most substitution by chestnut has occurred;
- identify the species, communities and habitats associated with these former woodland communities and their significance in relation to current biodiversity action plans and targets;
- assess the physical, biological and ecological impact of chestnut monocultures on ecological processes and biological communities associated with semi-natural, ancient woodland;
- investigate the effects of chestnut management regimes and silvicultural practices on wildlife potential; and
- consider the extent to which the influence of chestnut monocultures and mixed chestnut woodlands might be modified by appropriate 'restoration' management practices.

1.2 Scope

The study took the form of a desk review of literature, supplemented by a brief questionnaire survey of experts and scientists actively involved in woodland management and ecological research. Most information was obtained from published scientific accounts using cross-referencing, together with key word searches with appropriate search engines. This was supplemented by a one-day workshop held at the Wye campus of Imperial College on 20th February 2003, which included participants representing nature conservation and research organisations, woodland owners and the coppice industry (Appendix 1). Under the title *The honorary native: the significance of sweet chestnut in woodland conservation management*, the day's proceedings dealt with a range of issues, outlined below, that were valuable in formulating the questions and ideas for this review:

Does chestnut really deserve its 'honorary native' reputation? As a species still exploited for coppice markets, its value for butterflies, migrant birds and ephemeral ground flora is well established with conservationists. But what sort of woodland has chestnut replaced, and was that a more diverse' habitat for wildlife? If coppice markets remain uncertain, is there still a case for keeping chestnut woods? Would they survive non-intervention policies, or can they be successfully integrated into other forms of silviculture?

In the interests of nature conservation, should we continue to coppice uneconomic chestnut woodlands? Is it desirable or even possible to restore chestnut plantations to near-native woodland on ancient woodland sites? What practical habitat restoration solutions would we like to see, and what form should future management take?

2. History of sweet chestnut in the UK

2.1 Introduction of sweet chestnut into Britain

Sweet chestnut *Castanea sativa* is an introduced species that has been present in Britain since its introduction by the Romans (Bradshaw 2002). As an historic member of the British flora, Rackham (1980) considered the species an 'honorary native', capable of maintaining and perpetuating itself as a component of ancient, semi-natural woodland[2]. This was recently echoed in a survey report of ancient woodland sites on Forestry Enterprise holdings in England, in which sweet chestnut stands were described as

... part of the historic variation found within ancient woods. They have not been regarded as PAWS (Plantations on Ancient Woodland Sites) …. *given that there is no intention to change their character and composition other than to accept any native diversification that may arise naturally* (Spencer 2002).

Early writers, among them Evelyn in his work *Sylva, or a Discourse of Forest-trees* published in 1664, considered chestnut a native. According to Rackham (1980), the first writer to suggest that sweet chestnut might be introduced was Daines Barrington, a correspondent of Gilbert White in 1769. He was opposed by E. Hasted, the Kentish antiquary, A.C. Ducarel and J. Thorpe, who counter-argued that a species that readily regenerated from seed, occurred with stools commonly of varying size and distribution, and that sometimes grew intermixed with other tree species, was unlikely to be introduced.

Rackham (1980) summarised the evidence against chestnut as a native as follows:

- Archaeological evidence for chestnut first appears as wood and charcoal on Roman sites. Furthermore, evidence of chestnut timbers present in some medieval houses (suggested by Ducarel, c.1772) is unlikely as chestnut is commonly confused with oak timber.
- Chestnut is almost absent from the British pollen record. In contrast, chestnut pollen is found in quantity in some southern European deposits.
- Chestnut place-names can only be traced back as far as the 13th century, eg, Chest Wood near Colchester.
- The earliest written records are for the mid-twelfth century in the Forest of Dean. Fig. 2.1 summarises the locations of chestnut from early documents.
- Chestnut is a fast growing but long-lived tree. The Tortworth chestnut, 36 ft 1 in (11 m) in circumference in 1977 must date back to the Middle Ages, having been described as a tree of legendary antiquity in 1706. Eighteenth century writers have described other such old trees.

[2] ancient woodlands are sites that have been continuously wooded since at least AD 1600 (Spencer and Kirby, 1992)

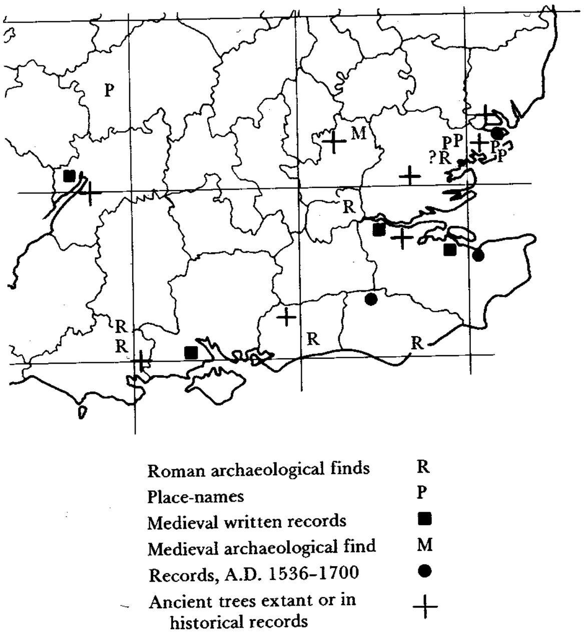

Figure 2.1 Records of chestnut, A.D. 0-1700, excluding planted trees (from Rackham (1980)

2.2 How much sweet chestnut was planted?

Drawing on accounts dating from 1772 by E. Hasted, A.C. Ducarel and J. Thorpe, Rackham (1980) concluded that a substantial proportion of sweet chestnut was not planted and had developed spontaneously. Points substantiating this view are that:

- chestnut grows freely from seed;
- individuals are not usually found in planted rows, or in regular stands;
- stools were, commonly, not all of the same age;
- some stools were very large;
- stools sometimes grew 'intermixed with other trees';
- some stands merged gradually and irregularly into connecting woodland.

Chestnut stands may have developed in areas favourable to its establishment from seed that was deliberately sown, dropped, or discarded by people. The distribution of these 'naturalised' chestnut stands might therefore reflect the areas inhabited by people who ate the nuts. In contrast, many stands of chestnut are clearly plantations. Rackham (1980) cites the example of Nap Wood (East Sussex) where an obvious plantation of regularly arranged small chestnut stools ends at a boundary, contrasting with the larger, irregularly spaced stools interspersed with various species in other compartments.

Some detailed case histories of deliberate chestnut planting are available, such as the nineteenth century records of the Mereworth and Preston Hall estates in Kent. According to Rackham (1980), most chestnut plantations date from after 1850, when the underwood occasionally commanded good prices (eg £14.8 ha^{-1} in Kent in 1875). Evans (1992) suggested that substantial planting of sweet chestnut coppice took place slightly earlier, from the 1820s, becoming widespread 30-40 years later. Both authors report a general decline in the demand for coppice wood at the time, but sweet chestnut was exceptionally in demand for the hop industry because of its long, straight, durable poles.

Roberts (1999) put the expansion of the hop-growing industry in Kent at over a century, roughly between 1780 and 1880. Hop fields initially needed 5,000-9,000 poles per hectare, and from the 1800s onwards the demand had to be met either by planting chestnut into woodlands cleared for the purpose, or on poor quality, typically free-draining agricultural land. For example, Roberts considered that Kings Wood near Challock, Kent, a substantial forest tract covering 600 ha and today consisting of more than 50% chestnut coppice, had probably still not been converted to chestnut coppice by the end of the eighteenth century.

From the available evidence it seems that the deliberate planting of open grazing or arable land may have played a relatively minor role in the recent expansion of chestnut plantations, although more detailed historical studies are needed to firmly establish this point. Nevertheless there is little doubt that large tracts of former ancient woodland were comprehensively cleared and substituted with chestnut within the last 150-200 years, while other areas were sporadically replanted in piecemeal fashion, or were colonised through natural invasion from adjacent plantations.

Recent census data, albeit based on the tiny sample of chestnut recorded in the National Inventory of Woods and Trees (Forestry Commission 2000), gives some insight into the scale of this substitution. This shows that of 1,080 ha of chestnut woodland owned by the Forestry Commission in south-east England[3], about 86% was present on former ancient woodland sites (Forestry Commission, personal communication). Thus, while the 'honorary native' view of chestnut may apply properly to ancient stands in mixture with other tree species on historically authenticated sites, many recently planted monocultures of chestnut are technically not ancient. However, the location of these plantations on ancient woodland sites, together with their potential to naturalise with other native broadleaved species, may give them a claim to 'honorary' status in the very distant future.

2.3 Pre-twentieth century use and management of chestnut

Coppicing of sweet chestnut was described in the first century BC by the Roman writer, Columella, and it is likely that the Romans brought this practice with them (Rackham 1980). The use of underwood for fuel and a range of products such as posts and wattle work is well recorded in documents from the Middle Ages.

According to Ellenberg (1988) in southern, Central Europe the *Betula*-Q*uercus pubescens* forests were converted to chestnut coppices from Roman times up to the Middle Ages. Posts were used for vineyards and buildings; litter was raked up and used in animal houses; young shoots were cut for fodder to feed goats and cattle; and fruits were smoked and stored, for bread-making and feeding to pigs. In the late Middle Ages chestnut flour was used to make biscuits in the Cévennes and Corsica and a bread made of chestnut and pulses in both Italy and Aquitaine (Rietbergan 2001). These chestnuts were probably domesticated trees, grown within wood-pasture or other parts of the agricultural landscape.

[3] (Kent, Sussex, Surrey, Hampshire, Isle of Wight, Buckinghamshire, Oxfordshire and Berkshire)

In Britain, Rackham (1980) cited historical evidence that sweet chestnut was grown primarily for its nuts and timber, rather than underwood during this period:

- Medieval records mention that chestnut was grown for its nuts and timber, but rarely mention underwood;
- In 1255 and 1278, the Sittingbourne chestnuts were cited as a source of timber (Roberts 1999) and were used in the same region for roofing, ceilings and walls);
- In 1278, "100 robora of chestnut, probably pollard bollings," were sent from Sittingbourne to Dover castle for building works.

Most post-medieval references refer to the use of chestnut for wood, but sometimes also for timber and nuts. However, it is not until the nineteenth century that pole sale records for Essex and Kent suggest that chestnut was important or more highly valued for coppice than other species.

At the end of the 19th century the demand for chestnut fell as wire supports reduced the need for high densities of wooden poles (Evans 1992). However, sales were to some extent maintained by markets for fence palings, posts and rails. Twentieth century markets are considered in more detail in Section 5.0.

3. The chestnut habitat

3.1 Native distribution of chestnut

Although chestnut was present throughout north-western Europe during the Tertiary era, its post-glacial centre of origin is thought to be in south eastern Europe and Asia minor, where it survived in refugia during the Pleistocene (Villani and others 1994). The pollen record indicates a gradual westward and northward expansion of the species: by 5000 BP it was widespread in Greece and southern Italy, and had invaded Spain and southern France by 2000 BP, attaining its present-day distribution around 1000 BP. It seems likely that this expansion was assisted both by Neolithic deforestation and, latterly, by domestication during the expansion of the Roman Empire (Huntley and Birks 1983).

Studies of allozyme diversity in chestnut indicate a geographic east-west partitioning of genetic variation roughly parallel to the Apennines in Italy, with further differentiation between NW and NE Turkey/Italy (Pigliucci and others 1990a,b). Isolation of chestnut populations in eastern Turkey was reinforced by mountain barriers in northern, western and southern Anatolia and by poor communication between the Black Sea and the Mediterranean during interglacial periods. Villani and others (1994) found a decline in genetic variation from Turkey to Europe, with less divergence between French and Italian populations than between those in western and eastern Turkey. This adds weight to the hypothesis that the Ponto-Caucasian populations are close to the centre of origin of the species and thus have retained the highest levels of gene diversity. The results are also consistent with the palynological record in suggesting successive waves of colonisation of new areas in western Turkey, Italy and France.

The origin of chestnut occurs at the transition between the Euro-Siberian (temperate) and Mediterranean floristic zones. In the former zone, chestnut-dominated forests of the Caucasus region occur mainly on southern slopes along the Black Sea coast, with some outliers persisting on the northern side of the Caucasus range. The current area of chestnut stands here is estimated at about 50,000 ha, having declined by 30% since 1956 as a result of disease and logging (Pridnya and others 1996). Associates are typically oriental beech *Fagus orientalis*, hornbeam *Carpinus betulus*, oak *Quercus petraea*, aspen *Populus tremula*, oriental spruce *Picea orientalis* and fir *Abies nordmannii*. These vegetation types generally occur on brown forest soils derived from metamorphosed Jurassic shales, on the lower slopes of mountain valleys. Common undershrubs are *Rhododendron, Vaccinium* and *Corylus.*

Similar mixed broadleaved-conifer forests occur along the Eastern, Middle and Western Black Sea regions of Turkey (Kaya and Raynal 2001). Chestnut also occurs in broadleaved deciduous forests in the Marmara region, along both sides of the Bosporus, and in the humid regions of the Aegean mountains, the latter within the Mediterranean floristic zone.

3.2 Climatic tolerances

According to Ellenberg's classification of central European vegetation, chestnut is a constant of two main communities:

- V1 colline-submediterranean vegetation dominated by *Quercus pubescens*, with an annual average temperatures of 12-14^{o}C and 600-1200 mm annual precipitation.
- V2 colline to submontane-insubrian vegetation, with average annual temperatures of c.10-12^{o}C and 1300-2000 mm annual precipitation.

The latter compares with the Euro-Siberian floristic region of Eastern Turkey, where chestnut occurs at mean annual temperatures of 10-14^{o}C, falling to 6-10^{o}C at altitudes of 1000-2000 m, with average annual precipitation exceeding 1000 mm (Kaya and Raynal, 2001). Within its natural range in the Caucasus Biosphere Forest Preserve, chestnut stands occur in a belt ranging from 200-1000 m, with annual rainfall of 1000-1500 mm (Pridnya and others 1996). Ellenberg considered that chestnut was relatively insensitive to drought and although generally tolerant of winter frosts, it was occasionally damaged by late spring frosts (Ellenberg 1988). In a transect stretching from the coast to 1745m in the Appennines of Tyrrhenian, central Italy, Blasi and others (1999) demonstrated that chestnut woods occupied a substantial part of the transition between temperate and Mediterranean conditions. In the upper region chestnut associated with beech *Fagus sylvatic* but with Turkey (*Quercus cerris*) and holm oaks (*Q. ilex*) at lower altitudes. Within this climatic zone, annual rainfall was 960-1450mm, the mean annual temperature 11-14^{o}c, with a summer dry period lasting 1-3 months.

The climatic transition between the Mediterranean and Euro-Siberian regions appears to have resulted in two functionally different chestnut ecotypes: an eastern, mesic type and a Mediterranean, drought-adapted one. Physiological studies of progeny from six European populations of chestnut, occurring in regions of contrasting mean annual temperature and precipitation, have shown that water use efficiency (the ratio of plant carbon gain to water losses) varies considerably between populations (Lauteri and others 2002).

3.3 Ecological tolerance of chestnut in Britain

The relative ecological tolerances of a range of commercial tree species in Britain are given in the *Ecological Site Classification* of Pyatt and others (2001). The most suitable conditions for chestnut production occur in the south of England, corresponding to relatively high seasonal accumulated temperatures (1475 - >1800 day degrees), moderate moisture deficits (>200 mm) and high continentality scores (Table 3.1). Soil optima cover a relatively narrow range, with moisture regimes ranging from fresh to moist, and nutrient regimes from medium to very rich. In all these respects, chestnut is most similar to pedunculate oak, cherry and lime in its requirements. It is classed as a relatively light demanding species, although it can tolerate shade in its early years. Natural regeneration is most likely on soils with a medium nutrient regime with moder or oligomull humus.

Table 3.1 'Suitable' and 'very suitable' ranges for sweet chestnut and other broadleaved, native trees, according to climatic, soil and indicator values comprising the Ecological Site Classification (from Pyatt and others 2001).

Species	**minimum accumulated temperature (day-degrees >5°C)**[1]	**moisture deficit range (mm)**[2]	**continentality (Conrad index)**[2]	**soil moisture regime**[1]	**soil nutrient regime**[1]	**shade tolerance (shade =1, full light =9)**
Castanea sativa	1200	60->200	7->9	v. moist -slightly dry	medium - v. rich	7-9
Quercus robur	1087	60-200	>9	wet - slightly dry	poor - v. rich	9
Carpinus betulus	1087	90->200	>9	wet - slightly dry	poor -v. rich	6-8
Quercus petraea	975	90->200	7->9	v. moist -moderately dry	poor -v. rich	8-9
Fraxinus excelsior	975	<20-200	<5->9	wet - slightly dry	medium –carbonate	8-9
Tilia cordata	870	90->200	>9	v. moist – slightly dry	medium - v. rich	7-9
Betula pendula	870	60-200	5->9	wet - moderately dry	v. poor - v. rich	8-9
Prunus avium	870	60->200	5->9	v. moist – slightly dry	medium - v. rich	6-9
Fagus sylvatica	775	60-200	5->9	moist -moderately dry	poor –carbonate	5-7

[1]values for the 'suitable' to 'very suitable' range of the species
[2] values for the 'very suitable' range of the species

The Forestry Commission survey of plantations of ancient woodland sites (Spencer 2002) classified them into broad ecological communities based on their understorey vegetation, following the British National Vegetation Classification (NVC: Rodwell 1991). The three main woodland vegetation types associated with chestnut in this survey were, following a gradient of increasing soil acidity, the ash-maple (W8), oak-bramble (W10) and oak-wavy hairgrass (W16) communities (Table 3.2). Samples comprising the National Vegetation Classification (Rodwell 1991) also included chestnut at low frequency (0-20%). They in turn are related to, respectively, the chestnut-lime, chestnut-hornbeam and chestnut-oak woods in eastern England described by Rackham (1980). Rackham's chestnut communities covered about 8% of the area of his sample of ancient woodland (*c* 7,000 ha) and were closest in their affinity to oak and hornbeam woods on acidic soils (Figure 3.1).

All three woodland types are common in the warm and dry, south-eastern lowland zone on soils ranging in reaction from acid to calcareous. Rackham (1980) refers to chestnut as a calcifuge species, associated with sandy or silty soils, but able to grow on a range of parent material. In southern England, most large-scale conversion to chestnut plantations appears to have occurred in W10 and W16 woodland communities. Generally the former occur on base-poor brown soils (pH 4-5.5) with variable textures, moisture regimes and humus types, derived from argillaceous rocks and superficial deposits. Typical examples are the Eocene London Clays and the clay-with flints over the North and South downs, where seasonal waterlogging results in gleying. More free-draining types with incipient podzolisation occur on Eocene sands and gravels in Essex and Kent, or sandstones in the Weald. Chestnut is most frequent on the moister soils within the *Anemone nemorosa* (10b) sub-community (Rodwell 1991).

Chestnut stands within the W16 community are more commonly associated with free-draining and sandy-textured soils in the south-east. Here the pH rarely exceeds 4 and the organic layer usually consists of mor humus. W16 types are common in the Weald on Cretaceous sands of the Tunbridge Wells and Folkestone and Hythe beds, and on the Eocene and Bagshot sands.

The acid soils of W10 and 16 contrast with those of the W8 community, which characteristically occurs on calcareous mull soils in the warmer and drier lowlands at pHs ranging from 4.5 to 7 or above. Soils are clays and clay-loams derived from soft, argillaceous parent material and are therefore prone to surface-water gleying. Chestnut here is most associated with the *Primula vulgaris-Glechoma hederacea* (W8a) and *Anemone nemorosa* (W8b) sub-communities, the former being more base-rich and with less tendency to gleying and associated with the Weald, Oxford and London clays.

The above range of site-types and soil conditions occupied by chestnut illustrate its wide tolerance. In eastern England, Rackham (1980) characterised the older, established chestnut populations in ancient woodlands as usually occurring on sandy or silty soils with a pH of <4, a mor humus litter layer and less than 20% clay. However, sweet chestnut can also grow well on waterlogged soils on heavy clays, especially in W10 woodland community types (Rodwell and Patterson 1994). Although there is a strong case that the realised niche of naturalised chestnut populations is centered on base-poor soils with reasonably good drainage during the growing season, further research is needed to establish this point.

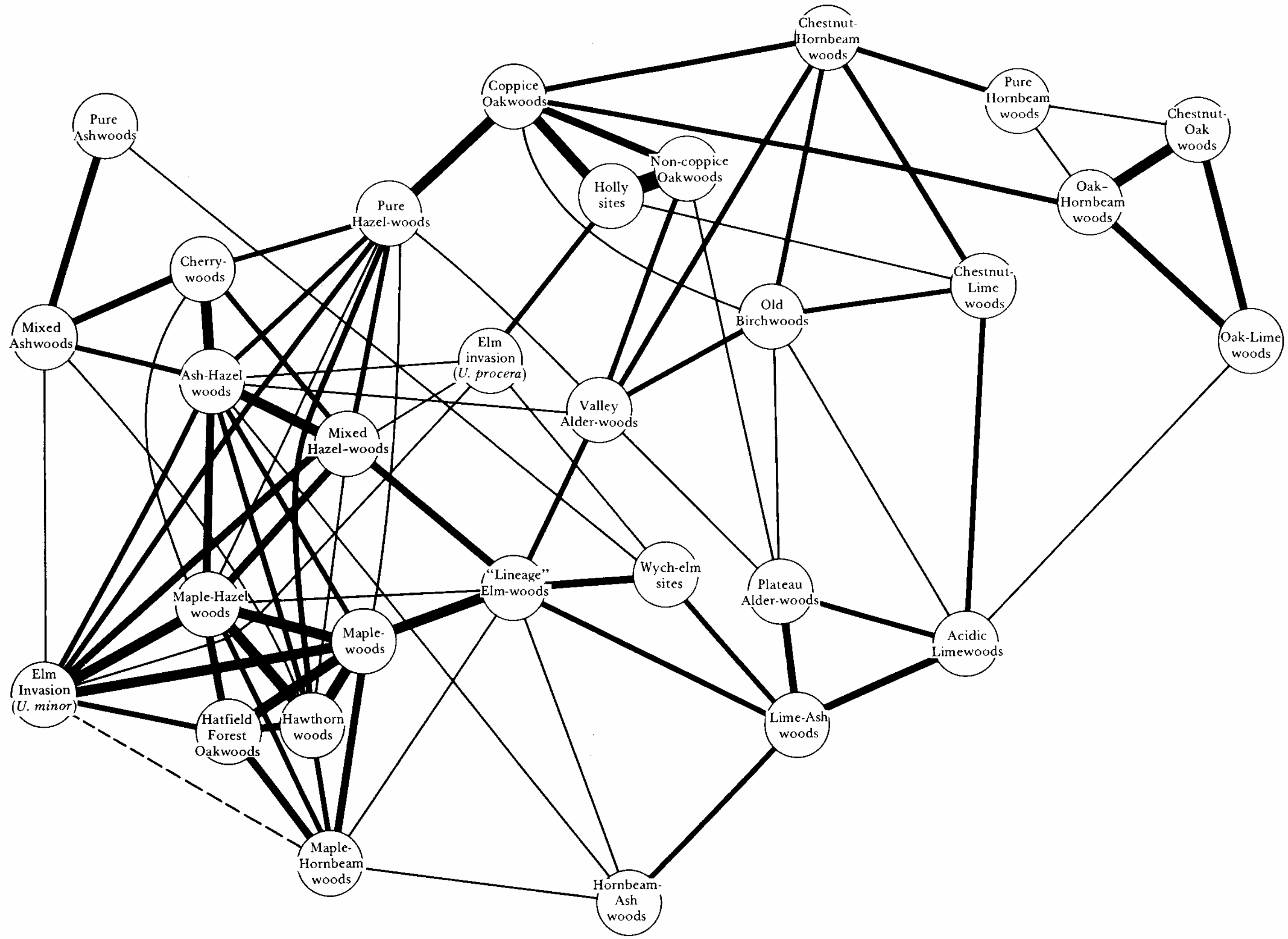

Figure 3.1 Affinities between tree communities in eastern England, based on their similarity and contiguity (from Rackham, 1980).

Table 3.2 British semi-natural woodland types in which *Castanea sativa* is commonly found. Tree, shrub and ground flora species shown are constants occurring across all sub-communities in each type, respectively (after Rodwell 1991).

Roman numerals refer to constancy classes (20% frequency) and figures in parenthesis to Domin (cover values).

Main woodland community	**W8** *Fraxinus excelsior-Acer campestre–Mercurialis perennis*	**W10** *Quercus robur-Pteridium aquilinum-Rubus fruticosus*	**W16** *Quercus-Betula-Deschampsia flexuosa*
Trees			
Castanea sativa	I (3-6)	I (1-10)	I (3-9)
Acer campestre	II (1-8)	I (1-4)	
Acer pseudoplatanus	II (1-10)	II (1-9)	I (1-4)
Alnus glutinosa	I (1-4)	I (1-9)	
Betula pendula	I (1-10)	II (1-10)	IV (1-10)
Betula pubescens	I (1-6)	I (1-9)	II (1-8)
Carpinus betulus	I (1-10)	I (1-9)	
Fagus sylvatica	I (1-8)	I (1-10)	I (1-7)
Fraxinus excelsior	IV (1-10)	II (1-8)	
Ilex aquifolium	I (1-8)	I (1-7)	I (1-4)
Larix spp.	I (1-8)	I (1-10)	
Malus sylvestris	I (1-3)	I (1-2)	
Pinus nigra		I (6-10)	
Pinus sylvestris		I (1-10)	I (1-8)
Populus tremula	I (3-8)	I (1-4)	I (3-4)
Prunus avium	I (1-5)	I (1-5)	
Prunus padus	I (1-6)		
Pseudostuga menziesii		I (6-10)	
Quercus hybrids	I (2-9)	I (1-10)	
Quercus petraea	I (1-9)	II (3-10)	II (1-10)
Quercus robur	III (1-10)	IV (1-10)	II (1-8)
Salix caprea	I (1-7)		
Salix cinerea	I (1-5)		
Sorbus aria	I (1-4)		I (3)
Sorbus aucuparia	I (2-5)	I (1-5)	II (2-4)
Taxus baccata	I (1-7)	I (1-9)	
Tilia cordata	I (1-10)	I (1-5)	
Tilia platyphllyos	I (4-8)		
Tilia vulgaris		I (3-7)	
Ulmus carpinifolia	I (3-10)		
Ulmus glabra	II (1-10)	I (1-7)	
Ulmus procera	I (2-7)		
Shrubs			
Cornus sanguinea	II (1-8)		
Corylus avellana	V (1-10)	III (1-10)	I (3-5)
Crataegus monogyna	III (1-7)	II (1-7)	I (1-2)
Crataegus laevigata	I (3-6)	I (2-4)	
Frangula alnus			I (3-4)
Euonymus europaeus	I (1-5)		
Prunus spinosa	I (1-8)	I (1-7)	
Rhamnus cathartica	I (1-6)		
Rhododendron ponticum		I (1-8)	I (1-4)

Main woodland community	**W8** *Fraxinus excelsior-Acer campestre–Mercurialis perennis*	**W10** *Quercus robur-Pteridium aquilinum-Rubus fruticosus*	**W16** *Quercus-Betula-Deschampsia flexuosa*
Sambucus nigra	II (1-7)	I (1-7)	
Viburnum lantana	I (1-8)	I (2-4)	
Viburnum opulus	I (1-5)	I (1-4)	
Ground flora			
Deschampsia flexuosa			V (1-9)
Eurynchium praelongum	IV (1-9)		
Lonicera periclymenum		IV (1-8)	
Mercurialis perennis	V (1-10)		
Pteridium aqulinum		IV (1-10)	IV (1-10)
Rubus fruticosus agg.	IV (1-10)	IV (1-10)	
Soil type associated with chestnut stands	base-rich mull soils (pH 4.5 to 7 or more): brown calcareous earths to rendzinas	brown earths of low base status pH 4 - 5.5	rankers, brown podzolic soils and podzols with mor humus
No. of species per sample mean and range	25 (5-64)	15 (1-39)	10 (3-29)

3.4 Plant species richness of the chestnut communities

Of the three main communities, W8 has the highest plant species richness, ranking in the top third of British woodland and scrub communities with a mean of 25 species per sample. By contrast both the W10 and W16 communities rank in the bottom third for species richness, with mean species numbers of 15 and 10 species per sample, respectively.

Given that much chestnut already occurs on acidic soils, the vegetation communities of such woodlands might be expected to be species-poor. Other major limitations to species diversity are likely to be the monoculture and dominance of the chestnut canopy in densely-stocked, managed plantations.

4. Range and extent of sweet chestnut in Britain

Chestnut is widely distributed in Britain, particularly in the south of the county, with scattered occurrences in Ireland (Figure 4.1). Although the method of presence and absence recording by the Atlas of the British and Irish Flora in 10km squares gives no guide to the abundance of the species, it is clear that upland areas and those containing little ancient woodland show fewer records. The Atlas has recorded a 59% increase in chestnut frequency since 1962, attributed mainly to improved recording between 1987 and 1999 and some increased planting (Preston and others 2002).

Figure 4.1 Distribution of chestnut in the British Isles in 1999 (Preston and others 2002). Reproduced with kind permission from Oxford University Press.

Woodland census data has been collected in Britain on eleven occasions between 1871 and 1999. Strict comparisons between census dates are hazardous because of differences in sampling intensity, changes in classification of forest type and the varying minimum woodland size limits used at each date. Thus, over the past fifty years, the total area of chestnut in Britain appears to have fluctuated between 19,000 and 30,000 ha. During this time chestnut growing as high forest has increased, while the area grown under standard trees (mostly oak) has declined. The overall area of chestnut coppice remained fairly constant at around 25,000 ha until the most recent census date of 1995-99 (Figure 4.2).

It is likely that these census data underestimate real areas, since chestnut often accompanies other species in mixed broadleaved high forest and coppice stands. These underestimates are evident when comprehensive habitat surveys at county level are compared with national census sample data. For example in Kent, the 1995-9 census recorded less than half the area of all chestnut categories, coppice, coppice-with-standards and high forest than that estimated from the Phase One habitat survey undertaken only a few years earlier (5,694 ha compared with 12,110 ha: in Government Office for the South East (2001) and Harvey (1996), respectively). Extrapolating from such observations, a maximum figure of *c* 40,000 ha of chestnut –dominated woodland in Britain seems not unreasonable[4].

[4] The 1979-82 census (Locke, 1987) suggested a total of 39,132 ha of pure coppice and coppice-with-standards, plus 71,900 ha stored (Evans 1984, 1992) = **111, 032** ha total, implying that only 35% of the coppice area was being worked at that time. Based on the proportion of chestnut within the worked coppice areas in England (51%), a crude estimate of the area of chestnut in British woodlands is therefore 111,032 x51/100 = **56,626** ha. However, this is certain to be a considerable overestimate since chestnut was then a commercially viable crop, in contrast to other coppice species, and a greater proportion of it was worked.

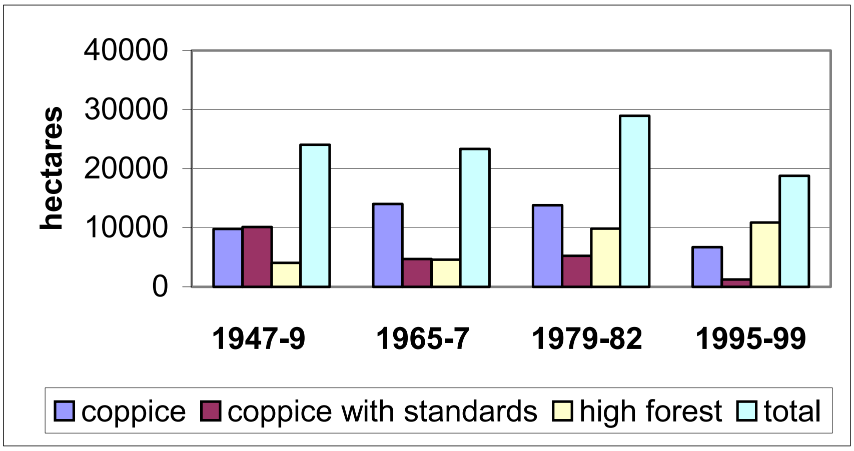

Figure 4.2 Sweet chestnut acreage in Britain, 1947-99, based on Forestry Commission census data.

By the 1995-9 census the *National Inventory of Woodland and Trees* gave the total chestnut area in Britain as only 18,788ha, of which 58% was classified as high forest (Forestry Commission 2000). This represents only 0.7% of the total forest area of Britain and 1.7% of the broadleaved woodland area. The vast majority of chestnut woodland occurred in England (96%), particularly in the south eastern counties of Kent and East and West Sussex (58%; Figure 4.3, Table 4.1). The apparent decline in area since the previous census can be explained by a number of factors (Braden and Russell 2001):

- a lower minimum woodland size was used previously, reducing the apparent area of woodland;
- the earlier census included chestnut coppice grown under standards which may subsequently have been classified by the canopy (usually oak) in 1995-9;
- much chestnut would have reverted to high forest as the markets for coppice products declined, being re-classified as mixed broadleaved high forest in the 1995-9 census.

Taking all factors together, there is no convincing evidence of a real decline in the chestnut acreage over the past 15-20 years, or indeed since the post-war census. Rather, the worked chestnut coppice areas have reduced significantly, gradually changing into mixed high forest stands as has happened in other parts of Europe. Over the same period, the area of chestnut high forest has remained relatively stable, but its proportion relative to the coppice area of the species has increased from 34% in 1979-82 to 57% in 1995-9. The coppice-with-standards proportion has also declined considerably, from 28% in England in 1979-82 to 15% in 1995-9, extrapolating from SE England census data. This suggests that the areas of chestnut remaining in active coppice management are increasingly simple coppice monocultures.

Table 4.1 Chestnut areas (ha) in South East England (Government Office for the South East, 2001).

County	Coppice	Coppice-with-standards	High forest	Total
Buckinghamshire	0	0	172	172
Oxfordshire	0	0	323	323
Berkshire	45	0	248	293
Kent	3922	611	1162	5695
Surrey	142	0	610	752
East Sussex	1147	394	1458	2999
West Sussex	632	50	597	1279
Hampshire	10	28	636	674
Isle of Wight	0	0	83	83
Total	**5898**	**1083**	**5289**	**12270ha**
%	48.1	8.8	43.1	100

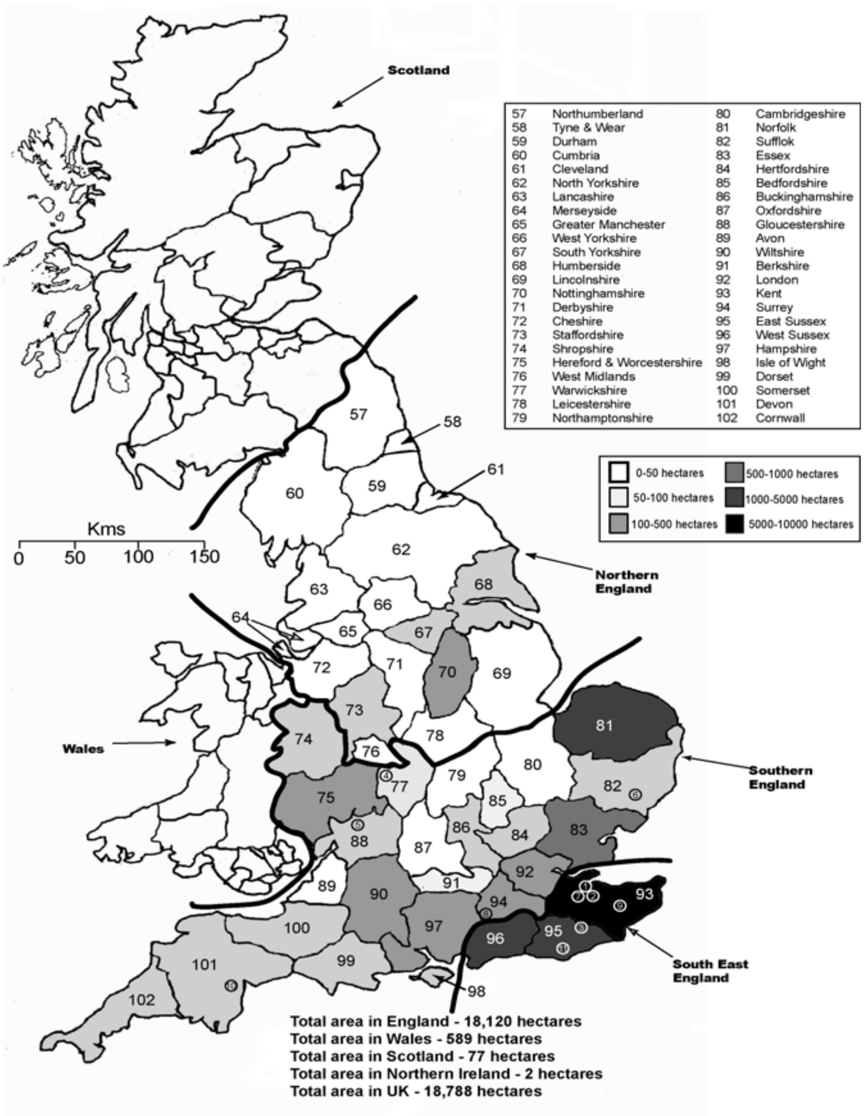

Figure 4.3 Sweet chestnut distribution in the UK, from Braden and Russell (2001). Reproduced with kind permission from Braden and Russell.

5. Silviculture and management

Guide to terminology

MAI - mean annual increment

dbh - diameter at breast height (normally considered to be 1.3 m)

$t\ ha^{-1}\ yr^{-1}$ - tonnes, per hectare, per year

$m^3 ha^{-1} yr^{-1}$ - cubic metres, per hectare, per year

5.1 Coppice management

Relatively little research has been carried out on sweet chestnut coppice. Most was begun by the Forestry Commission about 50 years ago, when seven experiments were initiated; one on chemical control, two on yield and four on silviculture. Nothing of value resulted from the four silvicultural experiments; three were closed and the fourth was damaged by squirrels (Harmer, personal communication).

Traditionally, sweet chestnut was grown as simple coppice or coppice-with-standards, in the latter case usually with oak. Simple chestnut coppice is cut on rotations appropriate to the product, site conditions, rate of growth and market demand. Compartments or coupes are typically monocultures of even-aged, single-storey crops with limited structural diversity. Standard trees introduce an additional canopy layer above the underwood, but their low timber quality and consequent marketing opportunities have allowed these ageing trees to dominate many coppice stands, once again reducing structural variation. Recruiting new standards and increasing the number of age cohorts within the standard tree population is thus a key management issue.

In both types of chestnut coppice system there are typically 800 to 1000 stools ha^{-1}, often cut on a rotation of 12-16 years, but this may vary depending on the market product required (see Section 5.2). The precise stocking density is less important for nature conservation purposes, but stool density is an important consideration for shade and ground flora management (Harmer and Howe 2003).

5.1.1 Establishment and restocking

Planting

Planting is regarded as the most successful method of commercial restocking. Everard and Christie (1995) recommend that plants are grown from local (UK) seed to reduce the risk of ink disease introduction: an example is the recent planting of locally collected nursery stock in the Forest of Dean. First year seedlings can produce robust plants 30 cm tall, growing to 50-60 cm two years after planting.

Vegetative propagation from stools

Sweet chestnut is suitable for layering as its shoots readily produce adventitious roots (Harmer and Howe 2003). In commercial plantations, shoot lengths are laid, partly buried and weighted with stones at positions where a new stool is required. This method requires specific expertise and careful weed control whilst the layered shoots establish, and is still current practice in some regions.

Natural regeneration

Natural regeneration is the preferred method of restocking semi-natural woodlands, but may not always be possible. Although chestnut produces copious mast in some years, there have been no detailed studies of its ability to naturally regenerate in the UK. Evans (1988) estimated the minimum seed-bearing age of chestnut at 30-40 years, with intervals of 1-4 years between large seed crops. While the nuts develop well at the centre of its distribution in southern England, chestnut suffers frequent predation of seed by small mammals and insects, as well as browsing of the young seedlings by deer (Section 10). Rackham (1980) considered that the nuts are less subject to predation than acorns and may germinate well, producing young seedlings in canopy gaps. In Alice Holt forest in Hampshire, Harmer (2003 workshop) observed that most nuts were attacked by maggots and that few seedlings were seen. In contrast, Everard and Christie (1995) considered that natural regeneration in the Forest of Dean was promising and further suggested that direct sowing could be made to supplement planted stock. Rackham (1980) also states that in some parts of England, for example Essex and Kent, "chestnut is capable of perpetuating itself indefinitely and probably of invading woodland of other species". It would seem that evidence for chestnut regeneration is at present limited, anecdotal and inconclusive.

5.1.2 Regrowth and cutting

Stool regrowth may be influenced by the age and size of the stool, seasonal effects and the height and method of cutting (Harmer and Howe 2003). Most guidance comes from practical, on-the-ground observations made by specialists in coppicing. Phillips (1971) found that cutting by chain-saw or hand axe made little difference to the length and quantity of the shoots produced in the following year, but considered that because saw cuts tended to separate the bark from the wood, these stools might be more liable to wind action and rot than those cut by hand axe. However, because of the small sample of stools (five treatment replicates), these results must be regarded as tentative.

When considering the age and size of stools in relation to regrowth it is rather difficult to separate their respective effect, as the stool size may or may not relate to its age. Harmer and Howe (2003) suggested that, in general, 'small' diameter stools produce relatively more shoots than 'large' diameter stools, while ageing stools produce fewer shoots. They do not suggest any hard and fast rules as to what constitutes a 'large' or 'small' stool, as this varies with the site. The age at which stools become moribund will also vary with the site, but stool death rates after coppicing are typically of the order of 4-5% (Evans 1984).

Further, the time of cutting has been shown to affect shoot regrowth. The optimum time is in the dormant season, between late autumn and early spring (Harmer and Howe 2003). This leads to better stool survival, greater numbers of new sprouts and stronger subsequent growth.

The height at which the stools are cut may influence shoot regrowth and stump longevity. Felling close to the ground is recommended as it encourages new shoots to grow at a lower level and to develop their own roots, and also reduces the risk of butt rot occurring in the stems (Harmer and Howe 2003).

5.2 Coppice yield and markets

Table 5.1 Sweet chestnut productivity in Kent (from Begley 1955)

Site	Age	Height (m)	Shoots stool^{-1}	Stools ha^{-1}	Stems ha^{-1}
Lyminge	17	7.9	8.2	820	6672
Challock A	16	9.1	6.5	687	4448
Challock B	16	10.4	6.4	425	2718
Challock C	16	10.7	5.5	971	5436

Some of the earliest yield data for chestnut coppice were collected by Begley (1955) in four stands at Challock and Lyminge in Kent (Table 5.1) and again by Ford and Newbould (1970), who measured just one stand at Ham Street Woods, also in Kent. Evans (1992) reported mean annual increments of c. 2-4 t ha^{-1}yr^{-1} (roughly the equivalent of m^3 ha^{-1} yr^{-1}) for most coppice species on conventional rotations. Rollinson and Evans (1987), investigating the yield of sweet chestnut coppice at twelve different sites in southern Britain, found little effect of site conditions on yield, but a productivity range of 4-12 m^3 ha^{-1}yr^{-1}, depending on age. At conventional coppicing age (15 years) the mean yield was 5.3 m^{3}ha^{-1}yr^{-1}, increasing to 13.5 m^{3}ha^{-1}yr^{-1} at 30 years. The former value increased to 8.7 m^{3}ha^{-1}yr^{-1} at 15 years if a minimum stem diameter at breast height (dbh) of 4 cm was adopted instead of the standard 7 cm dbh. These productivity data have been used by the Forestry Commission to estimate potential productivity of chestnut coppice crops in south-east England (Table 5.2).

Table 5.2 Potential annual yields of sweet chestnut coppice in south-east England (from Dannatt 1991)

County	Area (ha)	Yield (tonnes) 15 year rotation	Yield (tonnes) 20 year rotation
Kent	12,544	66,900	100,400
East Sussex	3,349	17,900	26,800
West Sussex	1,393	7,400	11,100
Total, SE England	17,286	92,200	138,300

Chestnut coppice products include: fencing, pales and rails, hop poles, posts, pulpwood, hurdles, trugs, bean and pea sticks, shingles, tent pegs, walking sticks and firewood. However, market demand for many of these items has declined or disappeared over the past few decades. Some examples of products produced from commercial chestnut coppice crops in the 1950s in Kent are shown in Table 5.3. The total quantity of products is given. The cleft pales were mainly 3 feet to 3 feet 6 inches long (0.91-1.06 m) and the posts ranged from 4 to 7 feet in length, but were mainly 4 feet 6 inches – 5 feet 6 inches (1.37-1.68 m).

Table 5.3 Total yield per hectare of sweet chestnut products from commercial coppice in Kent (from Begley 1955)

Site	Posts	Pales	Peasticks	Cordwood*
Lyminge	2706	22054	1853	18.0
Challock A	2283	34409	1915	16.6
Challock B	5901	26255	2409	12.8
Challock C	4149	66409	1421	17.5

*one cord consists of small diameter poles stacked in 4x8x4 foot clamps (1.02x2.03x1.02 m)

Markets for chestnut hop poles declined towards the end of the 19th and into the early 20th century, although poles were still widely used until the decline of the hop industry in the 1970s. This market loss has been partially met by the fencing market for posts, rails and palings. Typical products are given in Table 5.4.

Table 5.4 Chestnut coppice products

Coppice age (years)	Product
3-8	trellis materials
11 – 15	palings (high grade, straight poles)
25 – 40	post and rail crops
45+	post and rail and planking

Chestnut prices peaked in the 1980s at £2,500 ha^{-1} for the best paling stands, when the industry employed 1,500 people (Harmer and Howe 2003). Evans (1992) considered sweet chestnut to be the most important commercial coppice species worked at the time, but prices collapsed at the end of the 1980s and have not yet recovered to those of the 1970s (Harmer and Howe 2003). In the 1990s auction prices for standing chestnut in Kent were typically of the order of £650 ha^{-1}, until in 1999 declining sales forced abandonment of the auction system (Table 5.5).

Table 5.5 Summary of standing chestnut coppice prices sold through auction houses in Kent (from Bartlett 2003)

Year	Area sold (ha)	Price £ ha^{-1}	Total revenue (£)
1987	77.5	1,265	97,963
1990	64.7	805	52,058
1993	32.8	562	18,409
1996	27.2	588	19,006
1997	27.4	707	19,394
1998	9.4	712	6,708
1999	7.1	546	3,890

Current standing prices for quality chestnut paling crops are now less than £740 ha^{-1}, with up to a maximum of £1,235 ha^{-1} for post and rail crops (Kent County Council 2002). While prices for finished chestnut palings have tended to remain static and production costs have increased, the number of cutters and processors has declined. The result is a shortfall of supply, with the demand probably now double that of product availability (Bartlett, personal communication). There are similar problems with the supply of post and rail products, but information is scarce due to commercial confidentiality.

From the analysis above, it appears that much sweet chestnut coppice remains uncut because of limited supply chain capacity, which in turn restricts market development (Appendix 2). These trends are reinforced by census data showing an increase in chestnut high forest (Section 4) and Forestry Commission returns from the Woodland Grant Scheme, which over a six-year period between 1995 and 2001 recorded coppicing rates in woodland compartments in Kent amounting to 600 $ha^{-1}yr^{-1}$ for all coppice types (Coney, personal communication). Bartlett (2003) also carried out a pilot survey of coppicing activity in Kent over a four-year period between 1999/2000 and 2002/2003. In the first year this was based on returns from Parish Tree Wardens and other volunteers, but was later widened to include large landowners, managers and cutters. The sample showed that 30-60% of the coppice cut was of chestnut (Figure 5.1).

Assuming an annual cutting rate of c.167 ha of coppice per year for all coppice types between 2000/1 and 2003/3 and an average rotation age of 20 years, this could represent 167 x 20 = 3,340 ha operating within the coppice cycle, or 15% of the total coppice area (*c* 22,000 ha) estimated by the Phase 1 Habitat Survey (Kent County Council 1995). For pure chestnut, an annual cutting rate of 76 ha on a 20-year cycle gives 1520 ha, or 12-13% of the chestnut coppice area estimated by the Phase 1 survey. While the Kent Coppice Survey clearly underestimates coppicing activity from volunteer returns, the Woodland Grant Scheme will also omit commercial coppice-cutting which is not grant-aided. Despite the discrepancies between the two surveys, the overall impression is of a larger area of stored coppice converting to high forest, while smaller areas of semi-commercial woodland are maintained on rotation.

5.3 Storing coppice

By the early 1980s it had been estimated that 65% of all coppice crops had already been stored due to the collapse of former markets (Evans 1992). Deliberate storing of chestnut coppice is now being actively promoted, both to supply potentially high value markets for chestnut saw-logs (Everard and Christie 1995), mainly for export to Europe, and also to restore woodland to high forest for nature conservation purposes.

Stored chestnut coppice normally needs to be managed if the crop is to be marketed as high quality timber. Thinning to reduce stem number and remove inferior growth is recommended; singling allows selection of the straightest and most vigorous stem (Harmer and Howe 2003). Even if the purpose of storing is just for conservation, stored stools are more prone to windthrow and management for safety may be necessary.

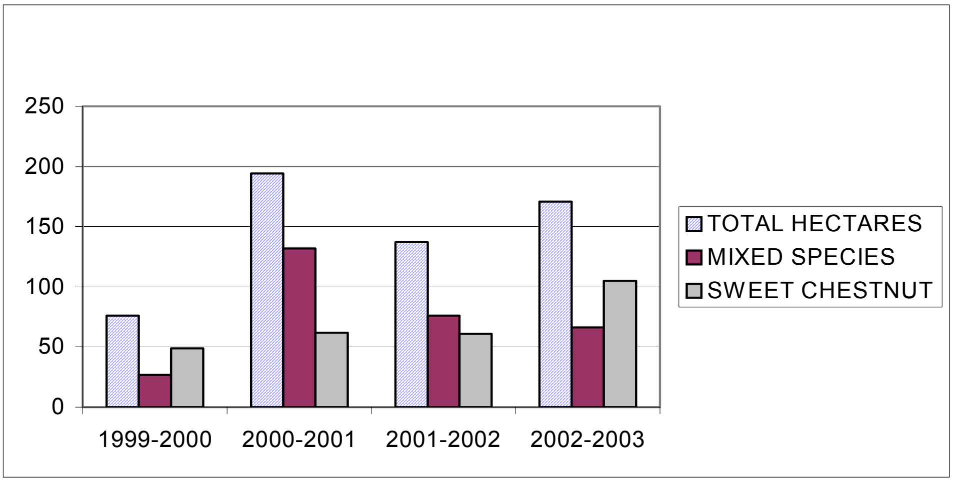

Figure 5.1 Results of the Kent Coppice Survey, showing annual cutting rates (ha yr^{-1}) of pure chestnut and mixed coppice crops (from Bartlett 2003)

Everard and Christie (1995) suggest the ideal age at which to store chestnut coppice by singling is between seven and 12 years. This avoids the regrowth catching up with the promoted stems and reduces the risk of crown damage caused by competition between stems. The same authors recommend keeping or introducing an understorey, when thinning chestnut stools, to reduce the growth of epicormic shoots. They quote the following advice:

1. Where the coppice growth is over 40 years old, or where the live crown is less than one third of the total tree height, the danger of blowing over or windthrow makes storing inadvisable.

2. Where coppice growth is 30-40 years old, aim for 200-250 ha^{-1} of crop trees and grow on a rotation of 40 years **plus** the age of the coppice.

3. For maiden stands or where coppice shoots are young and if the live crown is half the tree height, aim for 140-180 ha^{-1} of crop trees with a rotation of 50 years, or 50 years **plus** the age of the coppice.

5.4 High forest and timber

Although grown as timber in medieval times (Rackham 1980), today sweet chestnut is rarely grown as a timber tree in the UK because of the risk of ring 'shake', a separation of the wood between two annual rings, which renders the timber unusable (Rackham 1980; Everard and Christie 1995). Together with shake, fears of chestnut blight and ink disease may also play a part in landowners' reluctance to enter into long-term chestnut-growing (see Section 7.4).

Current markets for sweet chestnut timber include, saw-logs, veneer and planking. Everard and Christie (1995) have presented arguments - including yield results from stored coppice trials in south of England and the Forest of Dean – in favour of sweet chestnut as a timber tree.

These include:

- Quality and marketability of the wood.
- Hardwood sales of chestnut, even of modest size, command high prices in export markets and compare well with oak. For example, prices obtained at Rennies Hardwood Auction (Monmouth) in 1992 for oak standing timber of 21, 39, and 50 cm dbh were £9.94 m^{-3}, £5.93 m^{-3} and £44.23 m^{-3} respectively compared with chestnut standing timber of 17, 35 and 48 cm dbh at £10.39 m^{-3}, £41.67 m^{-3} and £18.27 m^{-3} respectively. Felled logs also commanded good prices.
- Losses resulting from shake can be reduced if the chestnut is managed on rotations of not more than 70 years: ink disease is mainly limited to wet soils
- The crop is relatively easy to establish and grows rapidly.
- Stored coppice crops in the south of England gave provisional yields (after gradual thinning) as follows: a mean annual increment (MAI) at 60 years of 8.8 $m^3ha^{-1}yr^{-1}$ (dominant stems of 46 cm dbh). The stocking density of the crop was c. 7,500 stems ha^{-1} at 10 years and 195 trees ha^{-1} by 60 years. For better sites in the Forest of Dean the equivalent figures were: an MAI at 40 years of 11 m^3 $ha^{-1}yr^{-1}$ (dominants of 41 cm dbh). The stocking densities were 6,000 stems ha^{-1} at 10 years and 250 stems ha^{-1} at 40 years.

5.5 Other markets

5.5.1 Nuts

Chestnuts have been utilised as a food source in Europe since prehistoric times, variously falling in and out of favour (Adua 1999). They have been eaten cooked or raw and used for flour, which is gluten free. Currently, nuts command relatively high prices in international markets with most export taking place from Italy, Portugal, Turkey and Spain to North American and Asian markets (Progetti 2003). Recently, the warmer summers in southern England have brought more regular crops of nuts (Everard and Christie 1995), but variation between seasons makes commercial viability unlikely at present. With projected global warming, chestnuts might become a crop for the future in southern Britain.

5.5.2 Edible fungi

According to Valjalo and Delmas (1982), sweet chestnut in France is associated with several mycorrhizal species, including desirable fungi such as *Amanita caesarea*, *Boletus edulis* and *Tuber melanosporum*. These three species have commercial value as edible mushrooms, but only *B. edulis* is native to the UK. As one of the best mushrooms and most widely sold *Boletes* spp., it is amongst the most important economic species collected in the wild (De Rougemont 1989). However, wild collection of *B. edulis* in the UK is still limited to fungal specialists and enthusiasts and it is not regarded as a commercial species. Its association with chestnut is not unique and alternative hosts include other members of the Fagaceae.

5.6 Potential markets

One important future use of chestnut may be for wood fuel. A study of the technical potential of wood fuel for renewal energy in Kent and East Sussex indicated that the maximum collectable biomass, (within a radius of 40km from existing chestnut coppice stands), was 10-15,000 oven dry tonnes per annum. Using gasification technology, chestnut coppice stands in Kent and East Sussex could supply around 50% of the fuel requirement of a 5MWe electricity plant or 25% of that of a 15MWe plant (Government Office for the South East 2001).

New technology, using finger-jointing and wet gluing of chestnut lengths sawn from small-diameter poles, is also being developed for laminate construction and joinery applications (Braden and Russell 2001).

Part II – Ecological effects of planting sweet chestnut

6. Vegetation

6.1 Plant species richness

In Section 3.3 it was pointed out that some semi-natural woodland communities are intrinsically richer in species than others, and that the majority of chestnut has been planted on acid soils. However, there has been little investigation of species richness in the understorey of different tree crops on comparable sites. One exception is the well-documented comparative studies of Ovington (1955), who examined ground flora development and changes in soil chemistry in unreplicated, 0.1ha plots of different broadleaved and conifer species. The original three sites were: West Tofts in the Breckland (planted 1930), Bedgebury, Kent (planted 1929-34) and Abbotswood in the Forest of Dean (planted 1905-6). Species at the latter two included *Quercus* and *Castanea* as well as the conifers *Pseudotsuga menziesii, Larix* spp., *Pinus nigra* and *Picea abies*. Both were old woodland sites on acid, brown earth soils and had inherited a complement of woodland flora, but at Bedgebury the *Castanea* plot was part of an old coppice shelterbelt, established *c*1817.

In 1952 the more mull-like soils under the broadleaved species had a richer ground flora than the rest. *Castanea* plots at both Abbotswood and Bedgebury had developed a similar understorey flora to that of *Quercus robur*, contrasting with those of the conifer plots. Twenty-two years later in 1974, Anderson (1979) repeated Ovington's recording of all three original sites. He found a general increase in the number of plant species per plot, attributed to thinning and the opening up of the canopy, together with a convergence to a more uniform number (about 20 per plot). As before, the chestnut ground flora was similar to the oak plot at Abbotswood. Anderson speculated that species abundance could be controlled by litter quality, thickness and structure, and that some plants could be inhibited by thick, loose-packed litter or by chemical conditions in these layers. He suggested that long leaves, for example chestnut and *Pinus nigra* needles, formed a loose mattress, causing the litter to dry out and leaving surface rooting plants and seedlings vulnerable to desiccation. This was less likely to occur under the short-leaved crops of oak and beech.

The effect of coppice management probably has the greatest single influence on the ground flora in the case of chestnut. In general, well-stocked, young chestnut stands recovering from coppicing rapidly form a dense canopy, reducing canopy transmission to less than 1% by the third season, levels at which only shade-tolerant species could survive (Mitchell 1992). Ford and Newbould (1977), working in pure chestnut coppice under standards at Ham Street, Kent, measured a logarithmic decline in field layer vegetation biomass in a chronosequence running from the second growing season to the end of the cycle (15 years). Species numbers peaked at five years but subsequently declined at 15 years. These results were similar those of Mason and Macdonald (2002) in a chestnut coppice woodland with oak standards, taken over a recording period of 11 years. In their study, incident radiation fell steadily after coppicing, reaching 1% of the ride values after 5-7 years. Species richness declined, but there was little effect of coppicing on the percentage frequency of *Anemone* (the ground flora dominant). Most other species (eg *Ranunculus ficaria, Adoxa moschatellina, Veronica montana, Euphorbia amygdaloides, Circaea lutetiana, Rubus fruticosus* and several grasses) peaked in abundance following coppicing, but had declined by the 4-5th year.

Considering the current decline of chestnut pole markets in Britain, the impact on plant species richness of storing coppice, and its eventual transformation to high forest, is highly relevant to this review. Unfortunately, no strict comparisons of high forest and coppice stands have looked into precisely this question. In one study in the Cévennes area of southern France, where downy oak *Quercus pubescens* has been replaced by chestnut, Gondard and others (2001) compared plant species richness along a successional gradient of unreplicated chestnut coppice stands and nut orchards. They found the greatest number of plant species in both operating and abandoned orchards (73 and 41 species $100m^{-2}$, respectively) compared with coppice which had lower species numbers (18-33 species $100m^{-2}$) (Figure 6.1). However, species richness in the managed orchard was largely an artifact of disturbance and cultivation, increasing therophyte abundance rather than true woodland species, whereas in the abandoned grove (75 years old) colonisation by shrubs and other plants following attacks of chestnut blight *Cryphonectria* increased species diversity.

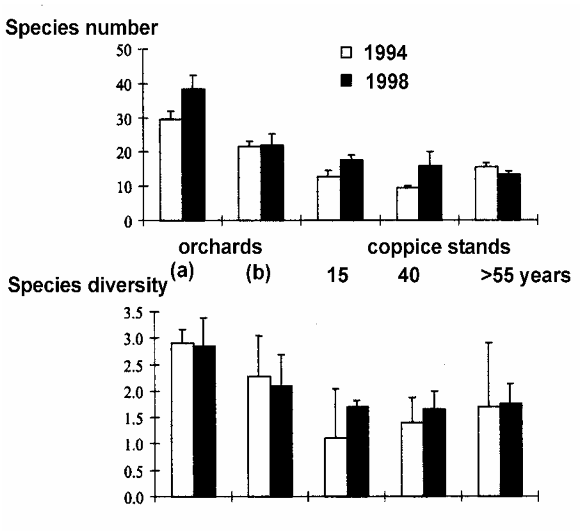

Figure 6.1 Plant species richness in (a) managed and (b) abandoned chestnut orchards and coppices, Cévennes, France (after Gondard and others 2001). Error bars show ± 95% confidence limits. Reproduced with kind permission from Kluwer Academic Publishers.

The general conclusion from the work cited is that regular cutting or disturbance in chestnut coppices does increase plant species richness, whereas vernal species and shade-tolerant ground flora dominants are relatively unaffected. Intensive management and high stool densities, on the other hand, discourage diversity through rapid canopy closure, a trend continuing for at least two-thirds of the rotation until the stands are re-coppiced. What is unclear is how diversity may be affected in chestnut-dominated stands which reach the high forest stage. The Bedgebury series suggests that these canopies may tend to support a ground flora similar to that under oak: however, one would anticipate greater heterogeneity to develop in time as the stem exclusion process and natural disturbance cycles begin to break up the dominance of the canopy.

6.2 Lichens

Each tree species has its own characteristic lichen flora (Broad 1989). Our native broadleaves, especially oak, support a good diversity of lichens (see Table 6.1). However, there are currently no records available for lichen numbers associated with sweet chestnut in the UK. In an Italian study, Loppi and others (1997) recorded 76 infrageneric lichen taxa in a sweet chestnut coppice woodland in Montieri, northern Tuscany. Although a direct comparison between the UK and Italian data is not possible, the figures suggest that the lichen flora associated with sweet chestnut may be less diverse than that of native species.

Table 6.1 The numbers of epiphytic lichens associated with key tree and shrub species of woodland types W8, W10 and W16 in Britain (after Broad 1989)

Key tree or shrub of woodland types W8, W10 and W16	Number of associated lichen species	Notes
Oak (penduncluate and sessile)	324	
Ash	255	Bark fissured and rather similar to oak but often of higher pH and therefore can support the *Lobarium* at a younger age
Beech	206	In spite of smooth bark, carries a flora very like oak in the New Forest, but has few epiphytes in chalk woodland or in the south-west. Bark of low pH
Hawthorn	No data	
Hazel	160	Quite rich especially in humid western areas
Downy and silver birch	126	Very acid bark
Sweet chestnut	No UK data 76 infrageneric taxa (Italy)*	*Records from site in Montieri in northern Tuscany, Italy (Loppi and others 1997)

Most sweet chestnut in the south-east is managed as coppice or coppice-with-standards. However, lichens are most abundant and diverse in environments that remain relatively stable for a long time (Broad 1989). Mature broadleaved woodland provides such a habitat with the bark of older trees supporting a greater diversity of lichens than the bark of younger trees. However, the regular cutting of coppice causes disturbance that is detrimental for lichens and the young bark of coppice poles supports only a limited range of lichens. Coppice-with-standards is more favourable than pure coppice as it offers continuity of large oak standards with the requisite mature bark.

A study by Rose (1974) on the lichen composition of oak woodland recorded the average number of taxa in 1 km^2 (or less) as follows:

Woodland type	Average number of lichen taxa in 1 km^2 (or less)
Ancient mixed oak forest	118
Mature oak clear felled and replanted during 18th and 19th century	50
Old coppice (species not given) with standards	42

The data indicate that although coppice-with-standards may be more suitable for lichens than pure coppice, mixed high forest provides by far the most suitable environment for lichens. According to Broad (1989) the disturbance of cutting restricts many lichens to the upper trunk and boughs of the standard trees in the coppice-with-standards system.

Evidence to date suggests that management of sweet chestnut for maximum lichen diversity is probably best met by allowing coppice stands to revert to high forest. Coppice-with-standards provides a compromise solution, but a continuous cover of mature broadleaved trees is essential for maximum lichen diversity. The data from Rose (1974) shows that the lichen flora in oak woods had not recovered 100 years after clear felling (see above). Broad (1989) recommends a shelterwood system involving the gradual removal of the tree crop in discrete groups and natural regeneration to replace the trees.

7. Fungi

According to Marren (2001) Britain has roughly 12,000 species of non-lichenised fungi and slime-moulds. This is not a precise figure as the list continues to grow, and the total may be nearer 20,000 species.

Fungi are key to the functioning of woodland ecosystems. Mycorrhizal fungi are mutualistically symbiotic with their host plants and play an important role in their nutrition, for example *Russula* species on beech, whilst pathogenic fungi (parasitically symbiotic fungi that damage their host) are key determinants of the composition and structure of woodlands. In addition, there are many organisms associated with fungi, in particular numerous invertebrates, whose numbers would be affected by the fungal composition. Any impact of sweet chestnut cultivation on fungal species diversity may therefore be ecologically significant. As the sweet chestnut coppice system allows for little accumulated dead wood, the diversity of saprotrophic fungi will also be less in coppices than in ‘natural’ woodland systems.

A number of fungal species are mentioned in UK Biodiversity Action Plans (Watling 1999) and the Sussex Rare Species Inventory List, 2002. UKBAP species associated with the woodland habitat in south-east England include: *Boletus regius* (Royal bolete), *B. satanas* (devil’s bolete), *Piptoporus quercinus* (oak polypore), *Hericium erinaceium* (hedgehog fungus) and the tooth fungi, *Hydnellum concrescens*, *H. scrobiculatum*, *H. spongiosipes*, *Phellodon confluens*, *P. malaleucus*, and *Sarcodon scabrosus*. The Sussex Rare Species Inventory list includes three additional species, *Hygrophorus nemoreus* (Basidiomycete fungus), *Pseudocraterellus sinuosus* (chanterelle) and *Russula lilacea* (Russula milk cap).

7.1 Fungal species diversity

The British Mycological Society (2003) has compiled a Database (BMSFRD) that currently contains over 600,000 records, including most macromycetes. This database was searched to obtain the numbers of fungal species recorded as associated with chestnut and the tree species of our target woodland types. The numbers quoted in the following discussion are a guide to the numbers and occurrence of the macromycete fungal species and may be regarded as representative, but in no way can they be considered definitive numbers.

The BMSFRD database records many more fungi associated with pedunculate oak Q*uercus robur* in the UK than sweet chestnut *Castanea sativa*, 1024 versus 578. Likewise there are more fungal species associated with other native trees, eg birch *Betula pendula*, sessile oak Q*uercus petraea*, ash *Fraxinus excelsior* and beech *Fagus sylvatica* (Table 7.1). However, some native species exhibited less fungal diversity than chestnut, including field maple *Acer campestre*, downy birch *Betula pubescens*, holly *Ilex aquifolium*, aspen *Populus tremula* and rowan *Sorbus aucuparia*. The other non-native, sycamore *Acer pseudoplatanus*, had more fungal associates than chestnut. (For the complete list of fungal species associated with chestnut see Appendix 3).

Table 7.1 Numbers of UK fungal species (mostly macromycetes) associated with tree and other species from the target NVC woodland communities compared with chestnut (after BMSFRD [online] 2003).

Reproduced with kind permission from the British Mycological Society.

	No of associated fungal species recorded in BMSFRD	**No of UKBAP fungal species**
Trees		
Acer campestre	243	
Acer pseudoplatanus	950	
Betula pendula	722	1
Betula pubescens	385	0
Castanea sativa	**578**	**6**
Fagus sylvatica	2402	7
Fraxinus excelsior	1096	2
Ilex aquifolium	365	
Populus tremula	204	
Quercus petraea	606	0
Quercus robur	1024	4
Sorbus aucuparia	207	
Shrubs		
Cornus sanguinea	34	
Corylus avellana	1317	2
Crataegus monogyna	445	1
Sambucus nigra	376	
Hedera helix	192	
Rubus fruticosus agg.	508	
Ground Flora		
Anemone nemorosa	36	
Deschampsia flexuosa	40	
Eurynchium praelongum	none	
Glechoma hederacea	14	
Holcus lanatus	60	
Lonicera periclymenum	48	
Mercurialis perennis	140	
Primula vulgaris	22	
Pteridium aqulinum	434	

7.2 Fungal species of conservation concern in south-east England

7.2.1 Diversity

The relative numbers of fungal species of conservation interest in the target woodland types of south-east England were obtained from the relevant UKBAP species list, the Local Biodiversity Action Plan (LBAP) species lists for Kent and Surrey, and the Sussex Rare Species Inventory. As the LBAPs for Kent and Surrey do not add to the UKBAP fungal species list, they will not be referred to further. Fungal species occurrence in south-east England was confirmed using *Fungi of south-east England* by Dennis (1995).

Cross-referencing the BMS database with the UKBAP list shows that there are two more species of fungi associated with chestnut (6) than pedunculate oak (4) (Table 7.2). In contrast, using the Sussex Rare Species Inventory, there are three more fungal species of concern listed for pedunculate oak than for chestnut. Overall, there seems little to choose between pedunculate oak, beech and chestnut in terms of rare species. Indeed, chestnut supports four UKBAP species of tooth fungi that have not yet been recorded on pedunculate oak, namely *Hydnellum scrobiculatum, Hydnellum spongiosipes*, *Phellodon confluens, Phellodon malaleucus* (Table 7.2). All these species are saprophytic and form ectomycorrhizal associations with trees (Table 7.4). Pedunculate oak is associated with four total species of concern (UKBAP and Sussex Inventory of Rare Species) not recorded for chestnut; two UKBAP species, *Boletus satanas* and *Piptoporus quercinus* and two further species from the Sussex Rare Species Inventory, namely, *Hygrophorus nemoreus* and *Russula lilacea*. In addition, the UKBAP species *Boletus regius* is known to associate with oak (although it is not included in the BMS database of oak fungal associates, presumably because of a lack of records), but has not been recorded in the UK on sweet chestnut. However, other species of *Boletus* are known to associate with sweet chestnut in Europe, for example *Boletus edulis* in France (Valjalo and Delmas 1982). All fungal species of concern associated with chestnut are found on one, or more, of the native tree species belonging to woodland NVC types, W8, W10 and W16.

Besides the number of species of fungi supported per tree species, it is also necessary to know how many rare fungi are supported by chestnut compared with the native woodland habitat. According to the BMS records, of the total thirteen fungal species of concern found in south-east woodlands (Table 7.2), twelve may potentially be supported by our woodland types, and all thirteen if *Boletus regius*, an associate of oak and beech, is included.

When beech is present in our target woodland types, the native trees may potentially support the full range of rare fungi associated with chestnut, plus five additional species. Beech is associated with six of the seven species of concern (UKBAP and Sussex Inventory of Rare Species) associated with chestnut, the exception being *Sarcodon scabrosus*, which associates with pedunculate oak and hazel. It seems therefore that chestnut does not contribute any unique fungal specialists to our lowland woodland types. The five species not supported by chestnut are the UKBAP species *Boletus satanus*, *Hericium erinaceium, Piptoporus quercinus* and the Sussex Rare Species Inventory Species, *Hygrophoreus nemoreus* and *Russula lilacea*.

The situation is somewhat different in native woodland types when beech is absent. Although the five UKBAP species not recorded on chestnut are still likely to be present, there are three tooth fungi recorded on chestnut that have not yet been found in association with pedunculate oak, hazel or any of the other main tree species of woodland types W8, W10 and W16. These are *Hydnellum scrobiculatum*, *Hydnellum spongiosipes* and *Phellodon mallaleucus*.

Table 7.2 UKBAP fungal species found in south-east England and their associations with key tree and shrub species of the woodland NVC types W8, W10, W16. Fungi of local importance are included. Fungal associations data collated from the British Mycological Soc Society on-line database (BMSFRD 2003).

Only presence is noted, see also key below. Reproduced with kind permission of the British Mycological Society.

Fungal Species	Common name	UK BAP	KENT	SURREY	SUSSEX	Key tree and shrub species of W8, W10, W16 and associated fungi								
		1	2	3	4	**SC**	A	Be	Hw	Hz	PO	SO	DB	SB
1. UKBAP														
Boletus regius	Royal bolete	x	x	x	x			1,4			1,4			
Boletus satanas	Devil's bolete	x	x		x			1,4			1,4			
Hericium erinaceium	Hedgehog fungus	x	x	x	x		1,4	1,4						
Hydnellum concrescens	Tooth fungus	x		x		**1**		1			1			
Hydnellum scrobiculatum	Tooth fungus	x		x		**1**		1						
Hydnellum spongiosipes	Tooth fungus	x		x		**1**		1						
Phellodon confluens	Tooth fungus	x				**1**		1						1
Phellodon malaleucus	Tooth fungus	x				**1**		1						
Piptoporus quercinus	Oak polypore	x		x	x						1,4			
Sarcodon scabrosus	Tooth fungus	x				**1**				1	1			
4. Sussex Rare Species Inventory species additions														
Hygrophorus nemoreus	Basidiomycete fungus				x			4	4	4	4			4
Pseudocraterellus sinuosus	Chanterelle				x	**4**	4	4		4		4		4
Russula lilacea	Russula milk cap				x						4	4		
UKBAP fungal numbers associated						**6**	1	7	0	1	4	0	0	1
Sussex Rare Species Inventory fungal numbers associated						**1**	2	4	1	2	4	2	0	2
TOTAL SPECIES OF CONCERN						**7**	**2**	**9**	**1**	**3**	**6**	**2**	**0**	**3**

KEY

SC = chestnut	A = ash	Be = beech
Hw = hawthorn	Hz = hazel	PO = pedunculate oak
SO = sessile oak	DB = downy birch	SB = silver birch

1. UKBAP
2. Kent BAPSG lists
3. Surrey woodland HAP
4. Sussex Rare Species Inventory.

1,4 = not listed as associated by the BMSFRD, but known to be associated with given species. Not included in the species counts.

Table 7.3 The number of British Mycological Society records (from BMSFRD [online], 2003) for the fungal species of concern that occur on chestnut.

These figures are only a rough guide to relative proportions. Pedunculate oak records are very under-represented as listings for mixed woodland, which probably include oak, have not been included. Only specific records of chestnut and pedunculate oak are shown. Reproduced with kind permission of the British Mycological Society.

Species	**Status**	**Total no of BMS UK records**	**SE counties – where species recorded on chestnut (No. of records for Kent, Surrey, Sussex)**	**No. of BMS UK records for pedunculate oak and chestnut**	
				PO	SC
Boletus regius	Endangered (Red Data List); BAP species	11	None	3	0
Boletus satanas	Rare (Red Data List ed 1), UKBAP	91	None	14	0
Hericium erinaceium	Vulnerable (Red Data List, ed. 1); BAP species	251	None	0	0
Hydnellum concrescens	Vulnerable (Red Data List, ed. 1); BAP species	244	Berkshire Kent (1)	67	8
Hydnellum scrobiculatum	Endangered (Red Data List, ed. 1); BAP species	137	Berkshire Surrey (5) West Kent (1)	10	14
Hydnellum spongiosipes	Rare (Red Data List, ed. 1); BAP species	130	Berkshire Surrey (4) Kent (1) Hampshire	42	11
Hygrophorus nemoreus	Vulnerable (Red Data List, ed. 1)	20	None	6	0
Phellodon confluens	Endangered (Red Data List, ed. 1); BAP species	57	Berkshire West Kent (4) Surrey (1)	17	11
Phellodon malaleucus	Vulnerable (Red Data List, ed. 1); BAP species	248	Surrey (2) Kent (1)	16	11
Piptoporus quercinus	UKBAP	35	None	33	0
Pseudocraterellus sinuosus	Vulnerable (Red Data List, ed. 1)	421	Kent (2)	9	2
Russula lilacea	Vulnerable (Red Data List, ed. 1)	7	None	3	0
Sarcodon scabrosus	Endangered (Red Data List, ed. 1); BAP species	156	Berkshire Buckinghamshire West Kent (3) Kent (1) Hampshire	9	3

7.2.2 Summary

Bearing in mind that the field survey records are not exhaustive, the BMS figures suggest that more species of fungi are associated with the dominant native tree species of W8, W10 and W16 woodland communities than with pure chestnut stands. Pedunculate oak, ash and silver birch all exhibit greater fungal diversity than chestnut. However, there is no evidence that chestnut coppice, when grown in mixed woodlands with natives species, would reduce fungal species diversity. Records show that similar numbers of fungi of conservation interest associate with oak and chestnut. Although greater numbers of these key species have been noted as present on pedunculate oak than chestnut (Table 7.3), this may be an artefact of the higher numbers of oak trees. Whatever the case, it seems that chestnut does not contribute significantly to fungal diversity or abundance. There are a couple of circumstances when these generalisations do not hold true. One species of conservation interest, *Sarcodon scabrosus* (Figure 7.1), appears to be particularly successful on chestnut. Further, it appears the rare tooth fungi are most likely to occur on either chestnut or beech, not on the other tree species of the target woodland groups. When beech is absent from a wood, chestnut may provide a good alternative for tooth fungi.

Rare fungi associated with woodland types, W8, W10 and W16 that are largely restricted to south-east England and as a consequence may be particularly affected by sweet chestnut cultivation in south-east England are: *Boletus regius*, *Boletus satanas*, *Hericium erinaceium* and *Piptoporus quercinus*.

7.3 Ecology

The numbers, relative numbers and range of fungal species are important for the functioning of the woodland ecosystem. The number of lignicolous, saprotrophic fungi has been shown to correlate with the amount of dead wood substrate available (Lagana and others 1999). In chestnut coppices, the absence of dead wood will greatly reduce opportunities for such fungi and hence the diversity of fungi. Increasing dead wood in the coppice system, for example by singling stools, extending rotations or encouraging standards over coppice will be necessary if these saprophytic fungi are to be kept in the system. One option suggested by the Tree Council (2003) is to plant fast-growing trees that will relatively quickly increase the amount of suitable decaying wood in the system. Planting sweet chestnut is recommended as it has similar wood-recycling fungal communities to oak, but is faster growing so will help bridge the age gap. Birch and willow are also recommended as they are short-lived and provide decaying wood on a relatively short rotation.

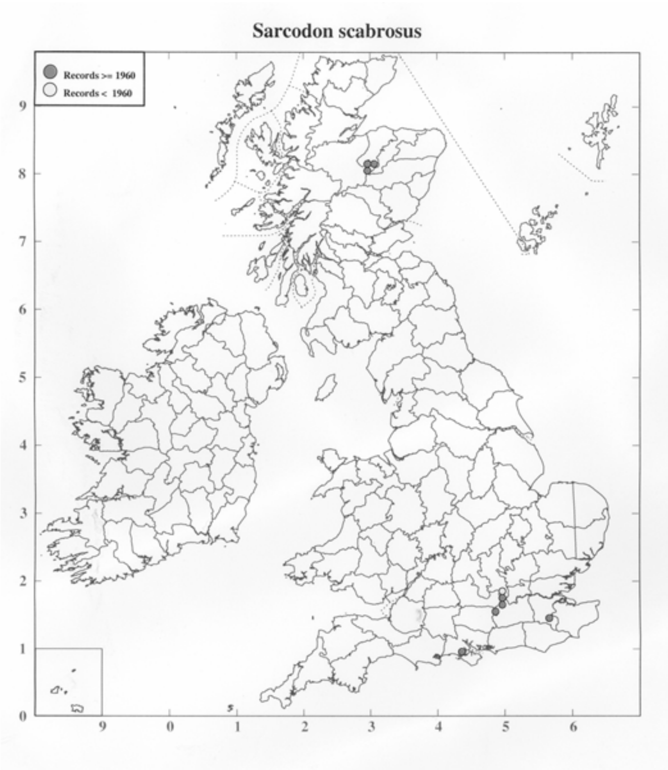

Figure 7.1 Distribution map of *Sarcodon scabrosus* in the UK (from BMS [online], 2003). With kind permission of the British Mycological Society.

Table 7.4 Fungal species of concern found in woodland types W8, W10 and W16 in south-east England and their role in the woodland ecosystem.

Species	**Role in ecosystem**	**Habitat** **Information provided by English Nature from http://www.arkive.org/ and the UKBAPs**
Boletus regius	Ectomycorrhizal associations with woody plants	Known mainly from grassy areas under broadleaved trees in ancient, deciduous woods, particularly hornbeam or beech woods but also oak, on calcareous or acidic sandy soils. Depends on old host trees. Many of its known host trees are old oak pollards.
Boletus satanas	Ectomycorrhizal associations with woody plants.	Seems to prefer calcareous soils. A southern species, it also likes warmth. Often found in association with beech or oak trees.
Hericium erinaceium	Hydnoid lignicolous saprophyte or facultative parasite.	Associated with old trees, particularly in woods where there has been a continuous history of old trees. Found on tree wounds, often high off the ground.
Hydnellum concrescens	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees.	Found in broadleaved woodlands, commons, and parks in association with oak, sweet chestnut, pine, larch and birch. It can be found on raised banks, stream banks, managed chestnut coppice and the sides of trackways. Shows a preference for open vegetation.

Species	Role in ecosystem	Habitat
		Information provided by English Nature from http://www.arkive.org/ and the UKBAPs
Hydnellum scrobiculatum	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees	Occurs under pine and broadleaved trees in woodland.
Hydnellum spongiosipes	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees	Occurs in well-established broadleaved woodlands, particularly oak, and is commonly found on stream banks, marl pits and banks with either bare or moss-covered ground, in acidic sandy soil, alkaline clays, and limestone.
Hygrophorus nemoreus	Mycorrhizal with forest trees	
Phellodon confluens	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees.	Broadleaved woodlands, but in Scotland it is also found in pine forests. It grows on sandy heathland, the sides of tracks, at the edges of marl-pits and on wood banks. It tends to be found in association with oak and sweet chestnut, and less frequently with silver birch, beech and pine.
Phellodon malaleucus	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees.	It occurs on sandy soils, typically on bare or mossy ground, and its distribution indicates that it prefers warm areas. Associated with a wide range of host trees including oak, sweet chestnut, birch, pine, and spruce.
Piptoporus quercinus	Saprophytic on angiosperm trees. Lives in the heartwood.	Found on the limbs and trunks of living or dead veteran oak trees (ie trees which are 250 - 800 years old), or on fallen heartwood.Typical habitats include medieval forests, deer parks, wood pasture and wooded commons.
Pseudocraterellus sinuosus	Saprophyte	
Russula lilacea	Mycorrhizal	
Sarcodon scabrosus	Stipitate hynoid saprophyte; forms ectomycorrhizal associations with trees	

7.4 Fungal pathogens

Fungal pathogens are part of the natural environment and also contribute to biodiversity. Natural mortality of trees varies, it regulates composition and structure within the woodland ecosystem, even influencing subsequent species that colonize the gap (Worral 2003). Understanding this complex system is essential for effective management of diversity. However, introduced diseases may not be considered 'natural' and can cause serious damage. Two such important fungal pathogens associated with sweet chestnut are chestnut blight *Cryphonectria parasitica* and ink-disease (*Phytopthora cambivora*, *P. cinnamomi*, *P. citricola* and *P. cactorum*).

7.4.1 Chestnut blight or chestnut bark cancer *Cryphonectria parasitica*

Chestnut blight is an Asiatic fungal disease that originated in China (Worrall 2003). It affects the shoots, branches and stems of sweet chestnut, forming cankers that grow inside the inner bark and cambium. A number of other tree species are also affected by chestnut blight. In a phytopathological inventory of mixed stands of sweet chestnut and other broad-leaved trees in Italy in 1994, *C. parasitica* was recorded on: oaks (*Quercus petraea*, *Q. pubescens*, *Q. robur*), hornbeam *Carpinus betulus*, ashes *Fraxinus* spp. and maples *Acer* spp. (Frigimelica and Faccoli 1999).

The susceptibility of tree species to this disease is variable. In its native Asia the fungus is a weak parasite. Chinese chestnut (*C. mollisima*) is resistant and the cankers that occur are of little consequence (Worral 2003), whereas the American chestnut *Castanea dentata* is highly susceptible and has been devastated since the fungus was introduced in 1876. Sweet chestnut is moderately susceptible, as are some oak species.

Chestnut blight is currently widespread in Europe and, although not yet present in the UK, it is progressing into Northern Europe. The disease affects chestnut forests and orchards in southern France (Guerin and others 2000) and Italy. In Germany it was first recorded in 1992 in Baden Wurttemburg (Seeman and others 2001). Sanitation fellings were carried out but were not successful in eradicating the disease. The Netherlands had an infection of the fungus in 1995 and again in 2001. Near its original distribution in the Caucasus, the disease was first observed in the 1930s (Pridnya and others 1996). In Croatia, chestnut blight has been present since the 1950s (Novak Agbaba 1999). Here the infections vary from moderate to severe, with disease being more intense in the Mediterranean regions of Croatia than the continental parts.

Although quarantine measures are in force it seems likely that chestnut blight will inevitably reach the UK. In terms of the health of sweet chestnut coppice, would the arrival of chestnut blight cause serious damage to the crop?

Rodriguez and Colinas (1999) state that *C. parasitica* is “presently the most important pathogen affecting *Castanea sativa* and *Castanea dentata*”. Novak Agbaba (1999) concurs with this assessment. But, according to Turchetti and others (1999), the level of chestnut blight damage in Europe is decreasing as a consequence of the development of hypovirulent strains, resulting from a viral hypoparasite that attacks the fungus (Anagnostakis 1999). Evidence for the effectiveness of “hypovirulence” comes from Switzerland where chestnut blight is largely confined to an area south of the Alps due to the less virulent strain (Heiniger and Rigling 1994). In Italy, control using the hypovirulent strain has led to a general recovery of the chestnut forests from chestnut blight (Anagnostakis 1999). In addition, chestnut breeding may develop blight-resistant varieties. The location and monitoring of diseases has also improved greatly with the use of aerial photography and GPS (global positioning system) to locate sites accurately prior to undertaking phytosanitary measures (Turchetti and others 1999).

Considering chestnut blight from another perspective, the low economic value of the crop may militate against campaigns of inoculation and phytosanitary treatments, as was seen in the Dutch ElmDisease epidemic of the 1970s. Furthermore, conservationists might welcome a non-interventionist approach to chestnut blight in the UK if it provided a natural means of diversifsifying chestnut monocultures, as has been observed in other parts of Europe.

However, the presence of a susceptible host species for chestnut blight in the UK could also increase the risk of it spreading to native tree species such as oak, hornbeam, ash and maple. Further research is required to answer these questions.

7.4.2 Chestnut ink-disease (*Phytopthora cambivora*, *P cinnamomi*, *P. citricola*, *P. cactorum*)

The *Phytopthora* species, collectively known as ink disease, are soil borne pathogenic fungi that generally cause root death. They are very common and widespread in Europe. *P. cambivora* and *P. cinnamomi* both infect sweet chestnut in the UK and are thought to have been introduced from outside Europe. Other less important species, *P. cactorum*, *P. citricola* and *P. gonapdyides*, have been identified in Italy (Vettraino 2001). In England, *Phytopthora* spp. are the most common cause of death in ornamental trees and shrubs (Buckzacki and Harris 1991). Ink disease is frequently observed on sweet chestnut coppice in old woodland (Buckzacki and Harris 1991), but currently *Phytopthora* species have only a small impact on British forestry (Brasier 1999).

Of the two major fungal diseases of chestnut in Europe, Turchetti and others (1999) considers that *Phytopthora cambivora* (ink disease) is now a more serious threat to chestnut cultivation than *C. parasitica* (chestnut blight). Anselmi and Vannini (undated) also describe ink disease as currently the most damaging disease of chestnut in Italy, with tree deaths occurring within two to three seasons. However, the relative importance of these diseases varies with location. There are no hypovirulent control measures for ink disease (*P. cambivora* and *P. cinnamomi*) and although it has been present in Europe for a long time, it has recently caused severe damage (Turchetti and others 1999).

The increasing impact of *Phytopthora* on forests and sweet chestnut in Europe has lead to growing concern for British forestry. Not only is the more susceptible sweet chestnut affected but also the oaks, *Quercus robur* and *Quercus petraea* have suffered serious decline in central Europe since the 1920s (Brasier 1999). *P. cambivora* has now been isolated as one cause of oak decline. According to Brasier (1999) "*P. cinnamomi* is most pathogenic at temperatures of 25^{o}C and above and does not survive freezing conditions in the soil". At present the UK climate is such that ink disease spread is probably limited, but projections suggest that it could become more widespread with an average warming of only 3^{o}C in future (Brasier and Scott 1994).

If ink disease becomes a more serious threat in the UK, the issue of how to control its spread should be reviewed. Portela and others (1999) have looked at soil factors and farming practice in relation to ink disease (*P. cinnamomi*) in chestnut groves in Portugal. No one factor was identified as responsible for the development of ink disease, although they confirmed that factors that damage the health of the tree lower the chance of its recovery. Chestnuts were more severely damaged on south facing slopes where they were exposed to higher radiation, higher soil temperatures and lower organic matter. Tree health was also adversely affected by shallow soil depth and frequent cultivation, both of which limited rooting. Trees with deeper more extensive roots fared better, as did trees on soils of higher fertility and higher organic matter. Calcium was reported as especially important for root health, whilst excess nitrogen was noted as detrimental.

Portela and others cite areas in Portugal where healthy chestnut groves grow adjacent to affected groves, although the climatic and lithological conditions are the same. They suggest that in affected areas it is the land management, site factors and their interaction that allows ink disease to take hold. It follows that management can help reduce ink disease incidence. Manuring to increase organic matter and soil fertility favours healthy trees, as does a closed chestnut canopy to keep the soil temperatures down. Liming raises calcium levels, aids root growth and thus helps control disease. In contrast, pruning increases the risk of ink disease damage as it allows greater exposure to sunlight and decreases the amount of litter fall. This might suggest that coppice is more susceptible to ink disease than forest trees.

Like chestnut blight, a future increase in ink disease on sweet chestnut needs to be considered in the context of the risk to our native trees. Diversification of stands due to chestnut mortality may provide a sink for the disease, thus encouraging its transmission to other species, such as oak. However, research by Maurel and others (2001) found that pedunculate oak (*Q. robur*) saplings artificially inoculated with *P. cinnamomi* had low susceptibility to root damage. Although at present ink disease does not pose much of a threat to our oaks, it may become important in future.

8. Invertebrates

Chestnut appears to have a reduced number of dependent insect species compared with native trees and shrubs present in the appropriate semi-natural woodland types. Southwood (1961) and Kennedy and Southwood (1984) demonstrated that the number of insect and mite species associated with different tree species was broadly related to a) the cumulative abundance of the tree through recent geological (Quaternary) history and b) the relative abundance of the tree species. Log-log transformations of insect numbers against tree abundance showed a linear relationship, indicating that the area occupied by the tree species in question was a key factor in explaining its insect diversity (Figure 8.1). Kennedy and Southwood's list for chestnut, a species occupying less than 1% of the British forest area, contained only 11 phytophagus insects: one Coleopteran, one Homopteran and nine species of micro-Lepidoptera, an impoverished fauna compared with the 400 species or more present on the more widespread *Quercus* and *Salix* species.

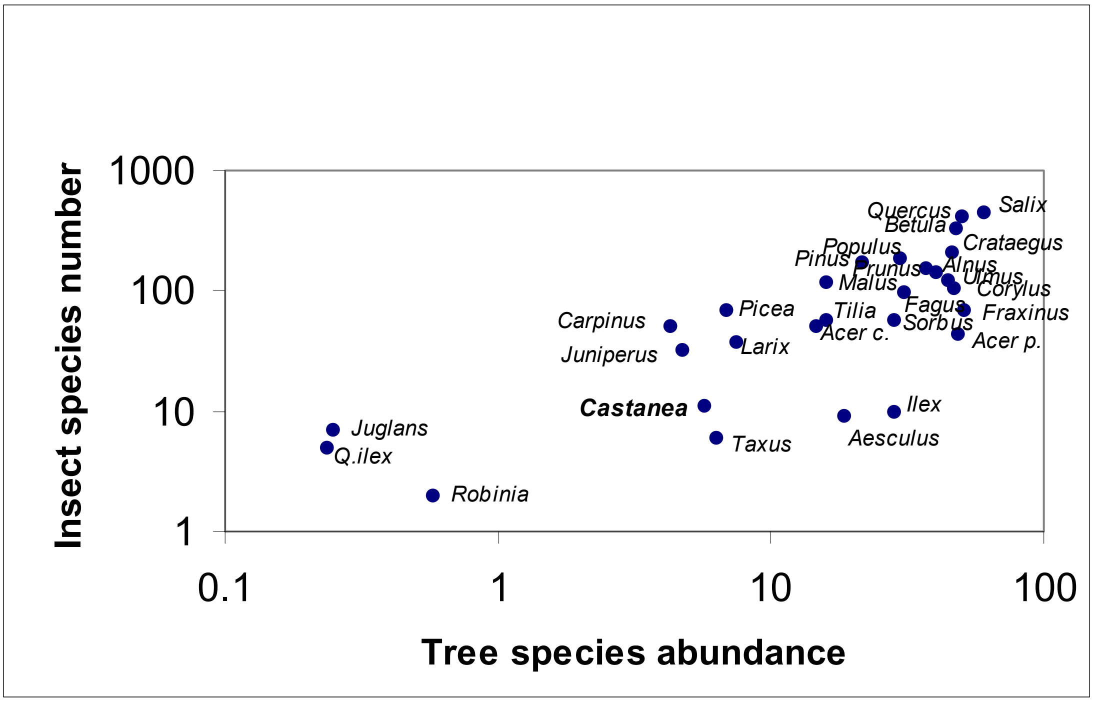

Figure 8.1 Relationship between insect species number and tree species abundance, measured in units of 1000, 10 x 10 km square x tetrad occupancy (after Southwood and Kennedy (1984))

Southwood's data were heavily dependent on the limited survey data available at the time and it could be anticipated that lists of phytophagus insects would expand with more systematic data recording. For example, later surveys of mature and coppice chestnut stands by Welch and Greatorex-Davies (1983) brought the total of Lepidoptera species on chestnut in southern Britain to 58, including 17 micro-Lepidoptera, of which six were leaf miners and three feeders on developing chestnut fruits.

Badmin (personal communication) listed 15 species, six Hemiptera and 11 Lepidoptera, as confirmed feeders on *Castanea* in the UK. Badmin also pointed out that large stands of chestnut were generally coppiced, and that this form of management could militate against insect species diversity. It was likely that mature trees would offer a greater variety of ecological niches, for example for insects attacking the developing flowers and nuts, and beetles attacking decaying timber, a possibility also considered by Welch and Greatorex-Davies (1983) in their study of mature and coppiced stands.

Many of the insect species associated with chestnut appear to be polyphagus and are not exclusive to the species. For example, only four species of scale insects have been recorded on chestnut in Britain, all of which are found on other tree associates (Table 8.1). A similar result was found by Vidano and Arzone (1987), who recorded the Typhlocybine fauna (Homoptera: Cicadellidae) of broadleaved trees and shrubs on Fagaceae (*Quercus, Castanea* and *Fagus*) in Italy. Of nineteen species verified as feeding on their hosts, *Castanea* hosted seven compared with 12 species for both *Quercus robur* and *Q. petraea*. The insect species most commonly co-hosted with those on chestnut were, however, the Mediterranean species *Q. cerris* and *Q. pubescens*.

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Table 8.1 Scale insect (Homoptera: Coccoidera) species feeding on chestnut and its tree associates in the UK (from ScaleNet [online], 2003).

Reproduced with kind permission from ScaleNet.

Family	**Species**	***Castanea sativa***	***Fraxinus excelsior***	***Quercus robur***	***Corylus avellana***	***Betula pendula***	***Betula pubescens***	***Quercus petraea***
Coccidae	*Ceroplastes ceriferus*					x		
Coccidae	*Eulecanium ciliatum*			x				
Coccidae	*Eulecanium douglasi*			x			x	
Coccidae	*Eulecanium tiliae*			x	x			
Coccidae	*Parafairmairia gracilis*		x					
Coccidae	*Parthenolecanium corni*	x	x		x			
Coccidae	*Parthenolecanium persicae*		x					
Coccidae	*Parthenolecanium rufulum*	x		x	x			x
Coccidae	*Pulvinaria vitis*				x	x	x	
Diaspididae	*Chionaspis salicis*		x					
Diaspididae	*Lepidosaphes conchiformis*	x	x		x			
Diaspididae	*Lepidosaphes ulmi*		x	x	x	x	x	
Diaspididae	*Pseudaulacaspis pentagona*		x					
Diaspididae	*Unaspis euonymi*		x					
Eriococcidae	*Pseudochermes fraxini*		x					
Kermesidae	*Kermes quercus*			x				
Kermesidae	*Kermes roboris*			x				
Pseudococcidae	*Phenacoccus aceris*	x	x	x	x	x	x	
	Total species number	**4**	**10**	**8**	**7**	**4**	**4**	**1**

8.1 Direct field comparisons of insect diversity

Few field investigations have compared the insect diversity of different tree species in the same locality or in comparable situations. In the Forest of Dean, Welch and Greatorex Davies (1983) sampled the larval Lepidoptera present on large, mature chestnut and sessile oak, when larvae of 24 species were recorded by beating the foliage on five occasions between May and September. The seven most common species on chestnut (accounting for

80% of the larvae collected) were also numerous on oak, but the total number of Lepidopteran species was only 21% of that on oak (Table 8.2).

Table 8.2 The seven most common Lepidopteran species on *Quercus petraea* and *Castanea sativa* in the Forest of Dean in 1980 (after Welch and Greatorex-Davies, 1983)

	Quercus petraea		***Castanea sativa***	
	larval no.	% total larvae	larval no.	% total larvae
Operophtera brumata	463	33.3	36	12.4
Agriopis aurantiaria	354	25.5	101	34.8
Tortrix viridana	258	18.6	20	6.9
Orthosia stabilis	28	1.9	38	13.1
Erannis defoliaria	24	1.7	8	2.8
Apocheima pilosaria	20	1.4	21	7.2
Cosmia trapezina	20	1.4	11	3.8
Total for 75 species	1167	83.9	235	81.0
Total larvae	1391		290	

In another direct comparative study, Hill and others (1990) examined the influence of coppice species and age on three major invertebrate groups – Dipteran flies, sap-feeding Hemiptera and Arachnidan predators. Coppice stands of 3, 8 and 12-year growth were compared in mixed chestnut-birch coppice at Church Woods, Blean, in Kent. Five sample trees were selected, spraying trees using a knock-down, synthetic pyrethroid insecticide in each period sampled. Although there was little effect of age, birch had consistently higher invertebrate densities and biomass than chestnut (Figure 8.2). Chestnut also had fewer invertebrates in a nearby mixed-species, mature coppice under oak standards than either hornbeam or hazel (Figure 8.3). Stand structure probably also had an effect, as the numbers of invertebrates on chestnut were lower in the coppice-with-standards plot.

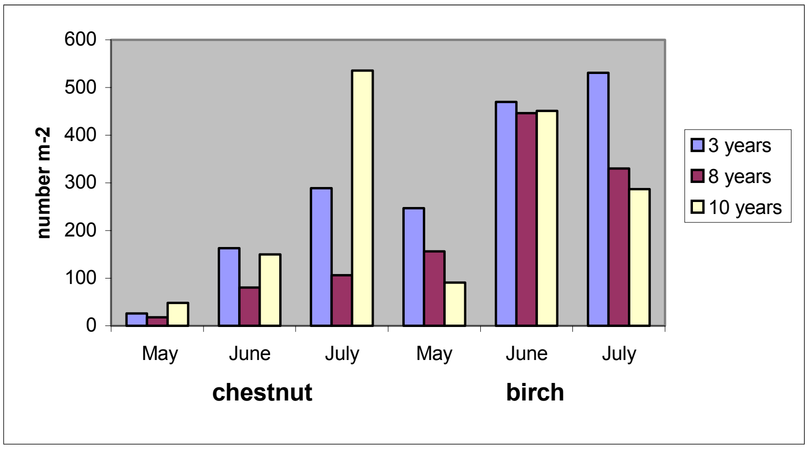


Figure 8.2 Invertebrate densities recovered from birch and chestnut coppice of different ages in Church Wood, Blean, Kent.

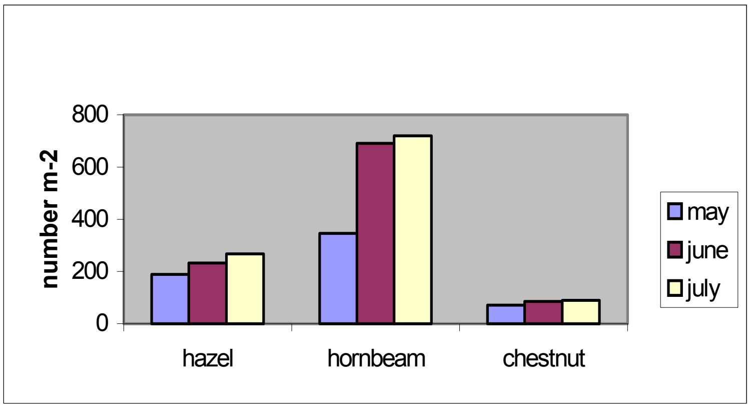


Figure 8.3 Invertebrate densities in different coppice species under oak standards in Church Woods, Blean, Kent.

Both comparable studies broadly support Southwood's original hypothesis in finding fewer insects on the introduced species. There remain some difficulties of interpretation, since chestnut growing in mixed stands with other tree species may derive some of its insect population from them. As Welch and Greatorex-Davies remark, such studies highlight the poor state of knowledge of insect fauna on introduced species, but they also indicate that *Castanea* has acquired a substantial insect fauna since Roman times, as predicted by Kennedy and Southwood.

8.2 Notable species

A highly localised species in southern Britain, the waved carpet moth *Hydrelia sylvata* uses a range of food plants in coppice woodland including alder, birch, sallow and blackthorn. However, larvae have recently been reported feeding on sweet chestnut in Blean Woods, Kent (JNCC 2001). As a species listed in the Biodiversity Action Plan for the UK and classified as nationally scarce, its association with actively coppiced chestnut areas is worth noting. In a recent study in chestnut coppice in Rewell Wood, West Sussex, adult moths appeared to be more common in coppice which had closed canopy (ie after 4-5 years' growth) and were strongly attracted to ride habitat, whereas younger coppice was avoided (Clarke 2003). However, few larvae were found during systematic searches of the foliage using the beating method. At the same site single occurrences of adults and larvae of another UK Biodiversity Action Plan species, the scarce merveille du jour *Moma alpinum* were found in mature chestnut coppice.

Several other Lepidoptera occurring in chestnut coppice are listed in Biodiversity Action Plans. One of the most notable is the heath fritillary *Mellicta athalia*, a Red Data Book species confined to habitats in southern England (Warren 1987c, Barnett and Warren 1995). In southeast England it is a woodland species dependent upon its food plant, the common cow-wheat *Melampyrum pratense*, which grows in recently coppiced or felled sites of acid W10 (oak-bracken-bramble) woodland. Here the presence of chestnut seems less important than the fact that it is coppiced regularly, creating suitable young growth structure and micro-climate for the insect and its host. A similar case can be made for the pearl-bordered fritillary (*Boloria euphrosyne*), a species that also benefits from coppice management.

8.3 Insect pests

Chestnut has a number of insect pests in Britain and Europe. Of the latter, both the chestnut weevil (*Curculio elephas*) and the late chestnut tortrix moth (*Cydia splendens*) are economically important as they attack the fruit: for example, in some years *C. elephas* can account for up to 90% of the nut produce in central Italy (Speranza 1999). Fruit predation must also have a significant influence on the success of natural regeneration in semi-natural woodlands. A 17-year study of chestnut patches in the Lyon area of France by Debouzie and others (2002) found that, in an 'average' year, between one quarter and one third of the fruit was predated by *Cydia* and *Curculio*, the former accounting for most damage in this case.

Other tortrid pests widespread in Europe include the early chestnut tortrix moth *Pammene fasciana*. The larvae cause the early fall and desiccation of the fruit, overwintering in the bark or on the trunk and emerge as adults in the summer. The intermediate tortrix moth *Cydia fagiglandana* has a similar distribution and has been noted as particularly damaging in Campania, Italy, where most nuts are attacked by this moth (Speranza 1999). Like many of the tortrid species, it is polyphagus, reported also on hazel, oak and beech.

The European shot-hole borer *Xyleborus dispar* also attacks chestnut and other forest species but for preference uses fruit trees (apple, pear, apricot) as its hosts. Adult females bore into the wood of healthy trees to lay their eggs from which the larvae develop, feeding on xylophagus fungi (eg *Ambrosia*) in the galleries.

Alford (1991) lists five insect pests present on chestnut in Britain (Table 8.3). All are present on other woody hosts, and none so far appear to have reached epidemic proportions in this country.

Table 8.3 Insect pests feeding on chestnut in Britain (from Alford 1991)

Family	Species	Common name	Alternative hosts
Callaphididae	*Myzocallis castanicola*	an aphid	oak
Attelabidae	*Attelabus nitens*	oak leaf roller weevil	oak, alder, hazel
Tischariidae	*Tischeria ekebladella*	leaf miner	oak
Gracillariidae	*Phyllonorycter messaniella*	Zeller's midget moth	beech, hornbeam, Holm oak
Notodontidae	*Phalera bucephala*	buff-tip moth	several woody broadleaved species

9. Birds

9.1 Bird species diversity

It is generally accepted that increasing structural complexity at both the whole-forest or the stand levels, provides a wider diversity of foraging and nesting sites and results in a greater bird species diversity than that found in more intensively managed systems. In the case of chestnut, the system in which the crop is grown (for example in orchards, as simple coppice or coppice-with-standards), as well as the length of the rotation, profoundly affects the forest structure. Within this framework, species diversity in the tree, shrub and the field layers also modify the niches available for different bird species. These structural and vegetational components interact not only with each other, but also with non-forest components in the wider fragmented landscapes, making it difficult to separate the key factors influencing bird distribution and abundance (Bellamy and others 2000).

A number of workers have investigated the effects of woodland age structure on bird communities. Fuller and Moreton's (1987) classic, ten-year study of breeding bird populations in pure chestnut coppice at Longbeech Wood, Kent, showed a typical response. In the early growth stages after coppicing, open-ground and migrant species such as tree pipit *Anthus trivialis*, whitethroat *Sylvia communis*, linnet *Carduelis cannabina* and yellowhammer *Emberiza citirinella* were common. Following canopy closure at around 7-8 years, species richness showed a sharp decline, coinciding with canopy closure. The balance then shifted towards resident species preferring more complex forest habitats such robin *Erithacus rubecula*, blackbird *Turdus merula* and great tit *Parus major* (Figure 9.1). The decline with increasing canopy closure was probably related to loss of nesting and foraging niches as the field layer vegetation disappeared (Fuller and others 1989).

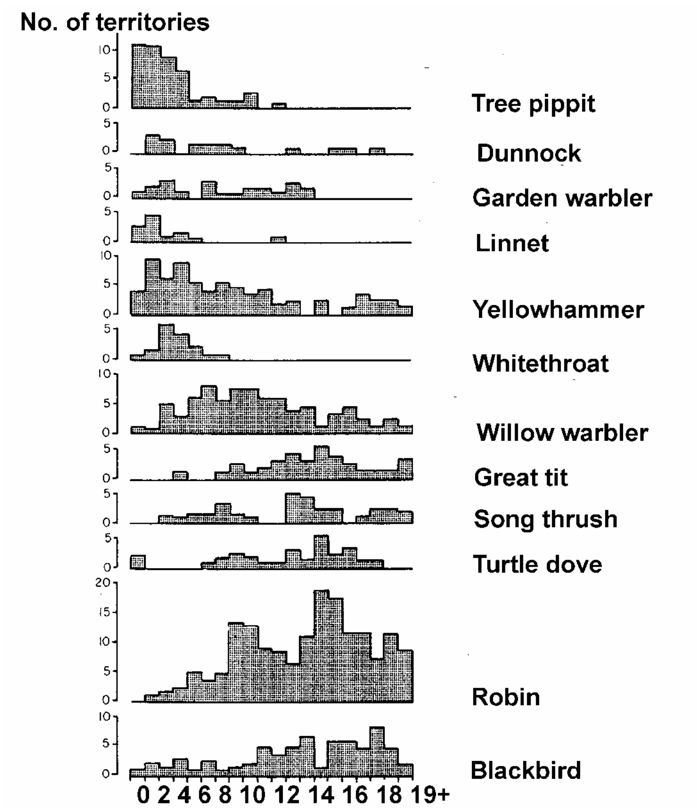


Figure 9.1 Population densities of bird species in relation to age of chestnut coppice in Longbeech wood, Kent (after Fuller and Moreton 1987).
Reproduced with kind permission from Blackwell Publishing

In another study at the nearby Ham Street Woods, Kent, Fuller and others (1989) demonstrated a similar preference of migrant birds for young coppice regrowth stages. Although the coppice in this case was predominantly hornbeam (only 18% was chestnut), the density of standard oak trees varied from 25 - >50 ha^{-1}. Although resident bird species appeared to be unaffected by standard tree densities, some migrants varied in their response, with nightingale *Luscinia megarynchos* increasing and whitethroat declining with increasing tree canopy cover. Deconchat and Balent (2001), in a study of fragmented chestnut coppice forests in southwest France, found that bird species richness was increased by the presence of standard trees. In this case species associated with forest interior conditions, large forest areas and long rotations such as nuthatch *Sitta europaea*, great spotted woodpecker *Dendrocopus major* and short-toed tree-creeper *Certhia brachydactyla*, were more likely to be present in coppice areas where standard trees were retained than in even-aged coppice patches.

Dechonchat and Balent's study also found that tree species richness *per se* had no evidant correlation with bird species richness. A similar conclusion was reached in a major study of breeding bird communities in Forest of Dean plantation crops that included chestnut, oak, beech, larch, Norway spruce, Douglas fir and Corsican pine (Donald and others 1998). Here no consistent differences in bird species richness or the proportion of migrants were found between broadleaved, coniferous or mixed stands, and there was no evidence that chestnut was any different in its bird-holding capacity to any of the other plantation species. The same study confirmed generic results in relation to age structure of the stands, but migrant birds, although common in the young growth stages, tended to re-emerge in older stands as the latter became structurally more diverse and open, with increasing amounts of low vegetation.

One recent study has identified bird species preferences for different vegetation types in the Val Surmassa Reserve in north-west Italy, a mosaic of coppice and coppice-with-standards woodland containing *Castanea sativa, Quercus petraea, Q. cerris, Q. pubescens, Q. robur* and *Robinia pseudoacacia* at different stages of growth (Laiolo 2002). Once again bird species richness and diversity were related to canopy height but also to canopy species richness, with hole-nesters, trunk and branch feeders and interior forest species most influenced by these factors. Canonical correspondence analysis further indicated that chaffinch *Fringilla coelebs*, woodpigeon *Columba palumbus,* blue tit *Parus coeruleus* and great-spotted woodpecker tended to be associated with oak or chestnut-rich plots, ie tree feeders or nesters.

Reviewing a number of studies on coppice sites in England, Fuller (1992) suggested several factors that could explain the relatively low abundance of bird species in pure chestnut coppice:

- the biomass of invertebrates is inherently low in chestnut stands compared with other broadleaved species within its adopted habitat (eg Hill and others 1990, Section 8). Insects are a major resource for birds foraging in woodland, especially summer migrants;
- the coppice structure produced by commercial management is generally simpler than that of mixed coppices. Chestnut is often grown in large coupes without standard trees, with little variation in the rotation length and thus no prospect of developing into more open, older stands;
- chestnut grows very rapidly from coppice stools compared with other species or naturally regenerated tree seedlings, casting a heavy shade. Canopy closure is also hastened by a managed high density of stools, causing impoverishment of the field and shrub layers;

- chestnut tends to be grown on acid soils, in woodland community types which are inherently infertile, restricting plant species richness and the number of niches available to bird species (Newton and others 1986).

In summary, many of the problems relating to chestnut as a habitat for birds are the result of growing it in monoculture on short rotations. At the same time, a by-product of this regularly coppiced structure is a high proportion of young growth stands, providing feeding and nesting habitat for a relatively high diversity of migrant species, many of which have restricted distributions in Britain, such as nightingale. However, specialists of high forest, including hole nesters, interior species and tree feeders clearly require much more complex and species-rich habitats and cannot be supported by intensive chestnut-growing.

9.2 Notable species

Bird species listed in the UK Biodiversity Action Plan that are relevant to the lowland, deciduous woods occupied by chestnut, include nightjar *Caprimulgus europaeus*, spotted flycatcher *Muscicapa striata*, and song thrush *Turdus philomelos*. Of these, the spotted flycatcher is more likely to thrive in coppice overstood by mature standards, while the nightjar thrives in the large coupes created in commercial chestnut coppice. Other notables listed in Local Biodiversity Action Plans for lowland deciduous woodland are hawfinch *Coccothraustes coccothraustes*, wood warbler *Phylloscopus sibilatrix*, redstart *Phoenicurus phoenicurus*, goshawk *Accipter gentilis* and nightingale *Luscinia megarhynchos*.

Nightingale has a south-eastern distribution in Britain, being most abundant in Suffolk, Sussex and Kent (Kent Biodiversity Action Plan Steering Group 1997). Nationally it is a declining species: species action plans generally recommend coppicing and scrub management on or adjacent to known sites, many of which contain a high proportion of chestnut coppice. Many of the other notable species are species of mature, open woodlands and are therefore less likely to be attracted to chestnut coppice unless mature, standard trees are present.

10. Mammals

Mammals using the woodland habitat in the south-east of England comprise: a) small mammals (four insectivores, five rodents (+ one non-native); b) predatory mammals (five carnivores), c) deer (two Artiodactyls + non-natives) and d) bats (12 Chiroptera).

10.1 Small mammals (rodents and insectivores)

Woodland structural diversity is particularly important for small mammal diversity. The early stages of the coppice system provide a ground flora and shrub layer that is key to the success of mice, voles and dormice. Standard trees in coppice add to both the structural and species diversity and thus to the range of small mammals that can be supported (Gurnell and others 1992).

A key species is the dormouse *Muscardinus avellanarius*, a UKBAP species named in the Lowland Mixed Deciduous Woodland HAP and protected under Annex IVa of the EC Directive, Schedule 5 of the WCA 1981 and Appendix 3 of the Bonn Convention. Its distribution corresponds closely with that of chestnut in the south of England (Figure 10.1).

Dormice require a shrub layer in order to move about above ground. According to Gurnell and others (1992), coppice stands younger than four years as well as older stands in which the shrub layer becomes shaded out, are both unsuitable for dormice. This supports earlier work by Morris and Whitbread (1986) showing a preference of dormice for species-rich coppice with interconnected trees and shrubs: they found that animals nested only in “young or relict coppice” and spent most of their time in mature coppice stands where there was good fruit production.

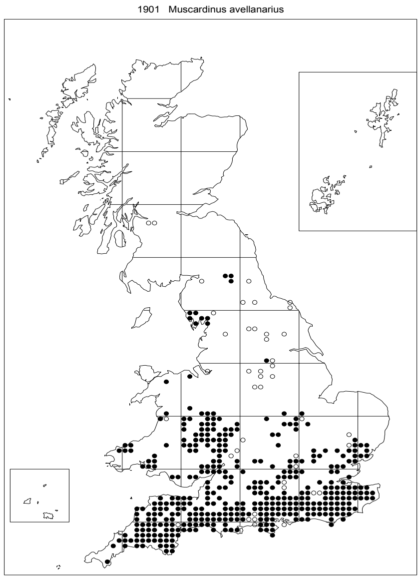


Figure 10.1 Distribution of the dormouse *Muscardinus avellanarius* in southern England.

(Map produced by the Biological Records Centre, CEH Monks Wood, using Dr Alan Morton's DMAP software, incorporating records supplied mainly by the Mammal Society. Kindly provided by Henry Arnold, 2004).

In chestnut monocultures, coppice stools are generally not interconnected by standard trees and their smooth, upright poles are less conducive to dormouse mobility compared with other species, such as hazel. Gurnell and others (1992) suggested that chestnut coppice was only likely to be suitable for dormice in mixture with other shrubs. They quoted two studies, not strictly equivalent sites, showing that many more dormice were captured in hazel coppice-with-standards than simple chestnut coppice (Table 10.1). The age of the coppice was also found to be important, with more animals trapped in 19 year-old chestnut coppice compared with younger stands.

Dormice require a continuous supply of flowers and fruits from April to November, which can only be provided by a diversity of woody plants (Bright and Morris 1989).

Table 10.1 Captures of dormice and other rodents in hazel *Corylus avellana* coppice with oak *Quercus* standards and in sweet chestnut *Castanea sativa* coppice of different ages (after Gurnell and others 1992.

Coppice age class (yrs)	**Number of shrub species**	**Number of trap-nights**	**Captures per 1000 trap-nights**			
Hazel coppice with oak standards (Isle of Wight, 1986)			dormouse	bank vole	wood mouse	yellow-necked mouse
5-7	8	320	52	175	0	-
25-30	8	3136	96	18	1	-
35-40	7	646	2	51	8	-
Sweet chestnut coppice (West Sussex, 1989)						
12	4	1600	6	2	1	2
12	4	960	1	0	0	0
14	1	640	0	0	0	0
15	1	640	0	0	0	0
19	7	1600	28	8	2	0

In spring flowers of hawthorn *Crataegus* spp., oak *Quercus* spp., honeysuckle *Lonicera periclymenum* and sweet chestnut all provide food. This is followed in late summer by soft fruits of bramble *Rubus* spp., yew *Taxus baccata* and wayfaring tree *Viburnum lantana*, with hazel *Corylus avellana* nuts becoming important later (Bright and Morris 1996). However, during midsummer food is in short supply, before fruits have ripened and after flowers have faded. Studies suggest that at this time dormice may rely on insects, especially aphids and lepidopteran larvae abundant on, for example oak, but in shorter supply on chestnut (Bright and Morris 1996).

In a study at Bradfield woods in Suffolk, Gurnell and others (1992) found that both the species diversity and density of mice, vole and shrew populations peaked in three-year-old coppice. With the notable exception of the yellow-necked mouse *Apodemus flavicolis*, most small mammals preferred young coppice. The most likely explanation for this preference was the presence of a dense ground flora in young coppice, this applies particularly to the grassland species such as field and harvest voles. With respect to the yellow-necked mouse, Marsh, Poulton and Harris (2003) found that its abundance was strongly related to the vertical forest structure, being positively correlated with woody climbers and standard trees, both of which aid vertical movement. They suggested that yellow-necked mice are more arboreal than wood mice *Apodemus sylvaticus* and are thus separated by a spatial niche.

Data from Gurnell and others (1992) imply that the abundance of bank voles and wood mice might be expected to be less under sweet chestnut coppice (Table 10.1). Taken with the evidence of sparse field layers and less diverse invertebrate populations under chestnut monocultures, it is reasonable to suppose that this habitat will be of lower quality for rodents and insectivore mammals than mixed-species coppices containing a variety of tree and shrub species.

10.1.1 Tree seed production and small mammals

Tree seed is an important food source for small mammals. However, the late fruiting time of chestnut has been cited as a factor that makes it a less favourable species for small mammals.

Both Evans (1984) and Gurnell (1993) found that hazel fruits earlier than chestnut. In his thirteen-year study at Alice Holt in Surrey, Gurnell found considerable variation in the time of seed fall between different years, but generally hazel fell first, followed by beech, sweet chestnut and oak. The fruiting periods of of the species were: oak (weeks 38-46), hazel (weeks 30-42), beech (weeks 34-44) and chestnut (weeks 32-46). For all tree species, there were weeks when up to 90% of the seed fall was consumed.

Canopy composition and the timing of seed fall may thus influence the relative numbers of small mammals by extending the period of productive foraging and allowing individuals to sufficiently increase fat stores for successful hibernation. However, in the case of dormice, there is no strong evidence that chestnut fruit is taken by the species. According to Bright and Morris (1993) hazel nuts are preferred when ripe and are particularly important for storing fat before hibernation. They further suggested that acorns might be of little food value for dormice as they are high in tannins (Bright and Morris 1996). It appears that continuity of food supply is the key factor, and for chestnut coppice to meet the food requirements of dormice it must be integrated with other woody species.

Gurnell (1993) gave the average calorific values of sweet chestnut, oak, beech and hazel fruits as, 19.4, 20.3, 29.4 and 34.5 kJ g^{-1} respectively. Although the poorest in terms of the energy provided, the quantity of chestnuts produced and consumed per unit area ultimately determines its importance as a food source. Gurnell's data shows that in some years sweet chestnut provided the most energy from seed fall, namely in 1975 and 1986 (Figure 10.2), even though the study site consisted mainly of oak standards over hazel coppice, with only 3% of the canopy occupied by chestnut. It follows that chestnut may be an important food source to small mammals when other species have poor mast years.

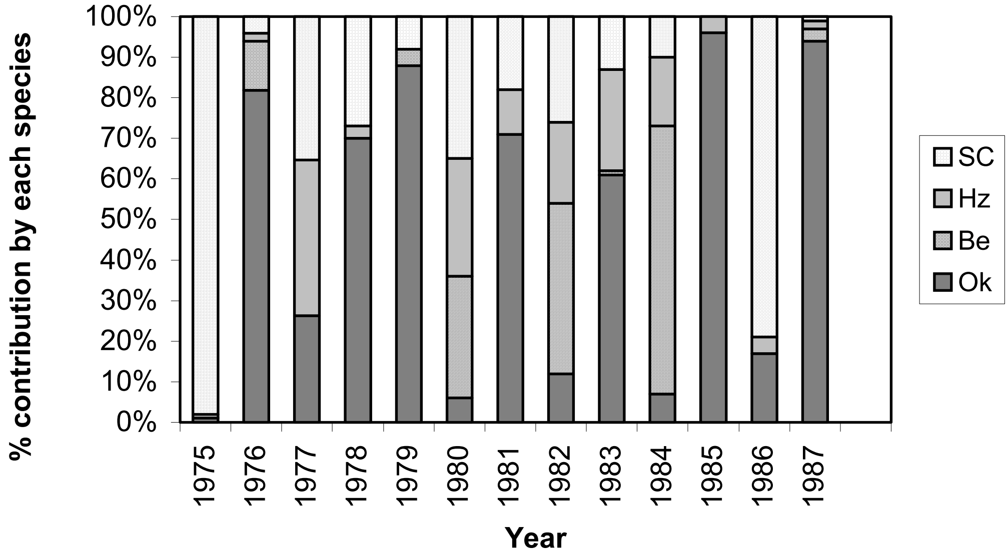


Figure 10.2 Percentage contribution to the total seed energy fall (MJ ha^{-1}) by oak Ok), beech (Be), hazel (Hz) and chestnut (SC) at Alice Holt, Surrey (after Gurnell 1993).

The more successful seed eating mammals may significantly affect the relative abundance of other seed eating species in the woodland habitat. For example, grey squirrels *Sciurus* *carolinensis* consume the majority of the hazel nuts by October (Gurnell and others 1992; Kenward and Holm 1989). This may adversely affect other species, for example the dormouse. The presence of standard trees in coppice not only benefit squirrels by providing seed, but also by providing nesting sites; stocking densities of about 25-50 ha^{-1} being optimum (Gurnell and others 1992). Although such densities are rarely found over worked chestnut coppices, chestnut itself, along with beech, sycamore and oak, is one of the species most frequently attacked by squirrels (Rowe and Gill 2000).

10.1.2 Summary: small mammals

Reviewing the evidence, it seems likely that sweet chestnut plantations are sub-optimal habitats for small mammals compared with coppice-with-standards containing a variety of trees and shrubs. To improve the woodland habitat for dormice, chestnut coppice cycles need to be long enough to provide a canopy, but should not exceed the stage when the canopy excludes other shrub species in mixed stands. The literature suggests that longer cycles than are currently worked are optimal (20 years or more: Gurnell and others 1992; Bright and Morris 1989), with a variety of growth stages present to provide greater structural diversity and a chance for the older coppice stands to fruit. The Kent Species Action Plan (Kent Biodiversity Action Plan Steering Group 1997) for dormice advises a long cutting cycle of 15-20 years in small coupes (less than 0.3 ha). Bright and Morris (1989) recommend small, irregular coupes of 0.2 ha or less, but not so small as to cause shading of adjacent coppice regrowth.

Management practices that favour dormice may not always provide the best conditions for other mammal species and other taxonomic groups. Increasing the coppice cycle length to favour dormice may conflict with species that require a short coppice cycle, eg the heath fritillary *Mellicta athalia* and nightingale *Luscinia megarhynchos* (Bright and Morris 1993). Further, expanding ride systems, adding paths, increasing ride width and clearing areas to provide conditions suitable for plant species of conservation interest will also have the effect of limiting the movement of dormice. Dormouse aids such as linking areas with hedges and mature trees and providing nestboxes are frequently suggested in the practical conservation literature (Kent SAP 1997; Morris, Bright and Woods 1989).

To achieve optimal small mammal diversity in chestnut coppice, a mosaic of coppice ages including young stands and stands of up to 30 years old would cater for both the ‘grassland‘ and ‘woodland’ species. Gurnell and others (1992) proposed that for a small wood (*c* 5 ha), retaining 30-50% of the coppice at 20-30 years old would meet the needs of the ‘woodland’ specialist species. Standard trees would add structural diversity and a habitat for tree dwelling species. Scallops or constrictions in the rides would enable above ground movement of the dormouse. For other small mammals, slightly larger coupe sizes of 0.5-1 ha, as part of an overall patchwork of stands, may provide a better habitat. This also fits Fuller’s (1992) criterion of providing large coupes to accommodate the territory sizes of summer-visiting birds.

10.2 Predatory mammals (Carnivores)

The four main native carnivorous mammals that frequent woodland in south-east England are the fox, stoat, weasel and badger. None of these are restricted to woodlands and all can travel some distance to find food and shelter. Numbers are inevitably affected by changes in the prey availability, and the amount that can be supported in a particular woodland habitat. As small mammal species generally do well in coppice-with-standards, the evidence presented here suggests that sweet chestnut coppice-with-standards managed as stands of different ages may also provide a viable habitat for carnivores, although not perhaps not as favourable as that of mixed broadleaved coppice.

Comparative evidence for diversity and individual numbers of small mammals in sweet chestnut coppice and native coppice is limited. No information on carnivores was located; further research is required.

10.3 Hoofed mammals (Artiodactyls)

10.3.1 Deer

Three species of deer are commonly found in woodland in south-east England, the native roe deer *Capreolus capreolus*, the introduced fallow *Dama dama* and muntjac deer *Muntiacus reevesi*. As large, free roaming animals, deer are able to locate food sources and suitable woodland areas relatively easily.

Ratcliffe (1992) reported that coppice habitat is suitable for deer, providing a source of food, shelter and cover. In the USA coppice has been used to increase deer densities, but rarely in the UK. Kay (1993) considered that “the recent promotion of traditional coppicing practices is likely to have played a role in encouraging the expansion of deer”. High numbers of deer are supported by the early growth stages (5-10 years) when there is much shrubby growth and dense ground vegetation (Ratcliffe 1992).

Maintaining deer populations may not be a role of conservation management in woodlands, but the palatability of sweet chestnut is an important issue for foresters. Nutritionally, chestnut leaves meet the nitrogen requirements of deer, but because of their low digestibility and high fibre are considered mid to low forages (Gonzalez-Hernandez and Silva-Pando, 1999). In a study of 74 young farm woodland plantations in east Suffolk, Moore and others (1999) found a negative, although non-significant relationship between fallow deer damage and the abundance of chestnut. Fallow deer damage was significantly and positively correlated with cherry and rowan. The mean percentage terminal shoot damage figures across all sites for chestnut, oak, cherry and rowan were 5.7, 6.7, 38.4 and 24.5% respectively (Table 10.2). Mean percentage lateral shoot damage figures across all sites for chestnut, oak, cherry and rowan were, 25.1, 37.4, 72.5, 66.8%. This suggests that, given the availability of these other species, fallow deer do not favour sweet chestnut over oak or particularly cherry and rowan (Moore and others 1999; Moore and others 2000).

However, another study by Kay (1993) in southern England found that sweet chestnut was one of the most heavily browsed species in coppice woodland. Fifty-three ‘coppice-with-standards’ woodland blocks on ancient woodland sites were surveyed, of which 39 were visited mainly by roe deer and 10 by fallow. The blocks had all been recently coppiced and 78 of the 1080 stools sampled were sweet chestnut. Comparing mean percentage shoot damage for hazel with sweet chestnut, the level of damage across all blocks was greater for sweet chestnut (Table 10.2).

Kay also found that the degree of browsing depended on the species of deer. Sweet chestnut suffered more damage by fallow deer than roe deer with a mean percentage shoot damage of 76.2 at fallow sites compared with 46.3% at roe sites. Ash was similarly affected, with 71.3

and 23.2% damage at fallow and roe deer sites, respectively. Hazel and field maple suffered similar levels of damage from roe and fallow deer.

Table 10.2 Deer browsing damage suffered by different tree species (after (a) Moore and others 1999 and (b) Kay 1993)

Species	(a) Young farm plantations No. of trees sampled	No. of plantations (total 74)	Mean % shoot terminal damage (±SE)	(b) Coppice woodland No. of stools sampled (from 53 woodland blocks)	Mean % shoot terminal damage
Acer campestre (field maple)	56	16	32.8 (11.3)	48	39.1
Acer pseudoplatanus (sycamore)	71	5	0.0	18	45.0
Alnus glutinosa (alder)	70	9	15.6 (10.8)	38	1.2
Betula pubescens (birch)	-	-	-	139	49.5
Carpinus betulus (hornbeam)	-	-	-	9	81.7
***Castanea sativa* (sweet chestnut)**	**2631**	**55**	**5.7 (1.5)**	**78**	**57.4**
Corylus avellana (hazel)	-	-	-	571	40.7
Fraxinus excelsior (ash)	290	9	5.9 (4.8)	103	24.3
Malus sylvestris (crab apple)	82	15	31.0 (10.4)	-	-
Populus spp. (poplar)	81	4	0.0	-	-
Prunus avium (cherry)	800	50	38.4 (4.5)	-	-
Prunus spinosa (blackthorn	74	14	0.0	-	-
Quercus robur (English oak)	2783	57	6.7 (1.3)	-	-
Salix caprea (willow)	-	-	-	15	61.7
Sorbus aucuparia (rowan)	255	30	24.5 (6.2)	-	-
Tilia cordata (lime)	115	7	26.1 (10.4)	61	55.5

Research on sweet chestnut coppice as a habitat for deer is rather limited, with evidence from different authors somewhat conflicting. The degree to which it is browsed depends on the deer population density and available forage in the surrounding area. It seems reasonable to assume that chestnut coppice is a viable habitat for deer, but probably less suitable than other native coppice species. The rapid exclusion of the ground flora in chestnut monocultures will make conditions less favourable, especially for fallow deer, which grazes rather more than it browses (Kay 1993). Coppicing *per se* is more likely to benefit deer than long-rotation, high forest systems: a study in Central Spain by Mateous-Quesada and Carranza (2000) found higher proportions of breeding females of roe deer in forests dominated by chestnut than in forests dominated by the deciduous oak *Quercus pyrenaica*.

Considering the impact of the habitat from another perspective, problems with coppice regeneration as a result of deer browsing are widespread and increasing. Browsing not only reduces the coppice crop, but can also have a dramatic effect on the composition of the ground flora, invertebrates, birds and small mammals, reducing plant species richness and simplifying structural diversity (Ratcliffe 1992; Gill 2000). Ultimately the natural regeneration of the canopy may be prevented through the browsing of tree and shrub seedlings.

According to Gill (2000), the small coppice coupes recommended in conservation management plans for a number of key wildlife species are particularly vulnerable to deer. In the study by Kay (1993), roe deer were found to select areas with good cover and inflicted less damage on larger blocks with relatively less edge perimeter. Damage by fallow deer was not correlated with perimeter length. Kay recommended larger blocks with small perimeter:area ratios to reduce roe deer damage, while Gill (2000) recommended fencing small coppice coupes to avoid damage.

10.3.2 Wild boar

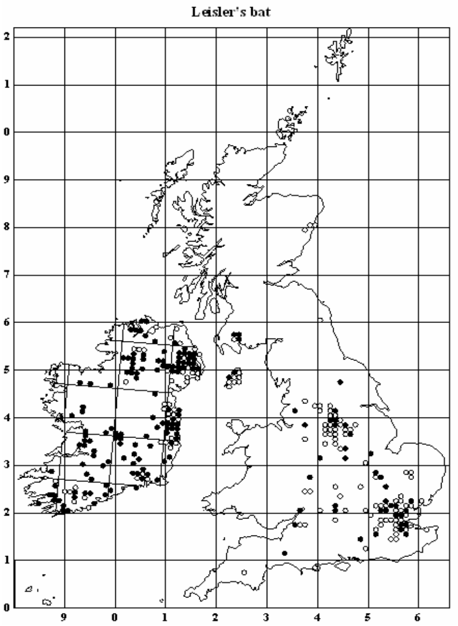
Wild boar *Sus scrofa* have been extinct from the UK since around the end of the sixteenth century (Howells and Edwards-Jones 1996), although on the Kent/East Sussex border there is a small population making use of coppice woodland. If reintroduced, as proposed by some conservationists (although not listed in the European Union 1992 Species and Habitat Directive as a species for which re-introduction should be considered), their impact on the tree seed crop could become a consideration. In the south-west Caucasus studies have shown that wild boar (along with other large mammals) consume 40-50% of the chestnut seed crop. To achieve seedling regeneration, it has been suggested that wild boar population densities should be no higher than 15-20 individuals per 1000 ha (Sokolov 1976).

10.4 Bats (Chiroptera)

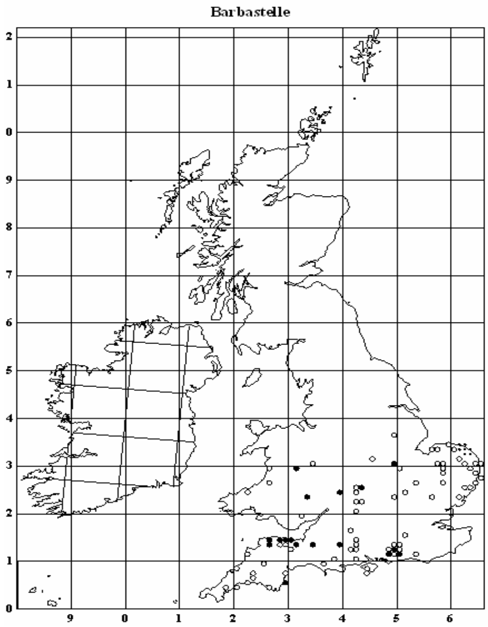
Of the fourteen native bats that frequent lowland broadleaved woodland in southern England, four species are relatively common (Pipistrelle *Pipistrellus pipistrellus* and *Pipistrellus pygmaeus*, Natterer's *Myotis nattereri*, Daubenton's *Myotis daubentonii* and brown long-eared bat *Plecotus auritus*); three species are considered vulnerable (Serotine *Eptesicus serotinus*, Leisler's *Nyctalus leisleri* and Noctule *Nyctalus noctula*) and two are endangered (whiskered *Myotis mystacinus* and Brandt's *Myotis brandtii*). There are a further four rare species: Barbastelle *Barbastella barbastellus*, Bechstein's bat *Myotis bechsteinii*, Nathusius's pipistrelle *Pipistrellus nathusii*, and Grey long-eared bat *Plecotus austriacus* (Figure 10.3). All of these forage for insects not only in woodland but at woodland edges, through open habitats and along water bodies. True woodland bat species are Natterer's, Leisler's, Noctule, Brandt's and Bechstein's, but even these range widely in open woodland and parkland. As chestnut plantations seem to provide a reduced insect biomass compared with mixed broadleaved woodland (Section 8), potentially they offer a poorer feeding habitat than mixed native woodland. However, if the chestnut stands are regularly coppiced, the provision of young growth and boundaries between adjoining compartments of different ages may produce more items of prey for bats than unmanaged woodland.

The relative importance of woodland as a habitat for bats varies with the species. The Noctule, Pipistrelle and usually the Bechstein's bats are tree dwellers in summer and winter, using tree holes and preferring mature trees (Morrison 1994). Other species, namely the Natterer's, Daubenton's, Leisler's, brown long-eared, grey long-eared and Barbastelle bats may roost in trees in the summer but dwell elsewhere in the winter, for example caves and buildings. The Serotine, whiskered and Brandt's bats are rarely found in trees in the summer. Mature and overmature chestnut trees could probably provide suitable sites for tree roosting bats and winter hibernation, but in practice such trees are rare. Chestnut coppice cannot provide such roosting sites, though coppice-with-standards systems may do so, augmented if necessary with bat boxes. The impact of both chestnut trees and coppice on bats has yet to be thoroughly researched. However, there are four bat species in south-east England that stand to benefit from the diversification of chestnut plantations: the vulnerable Leisler's bat and the rare, Barbastrelle, Bechstein's and grey long-eared bats.

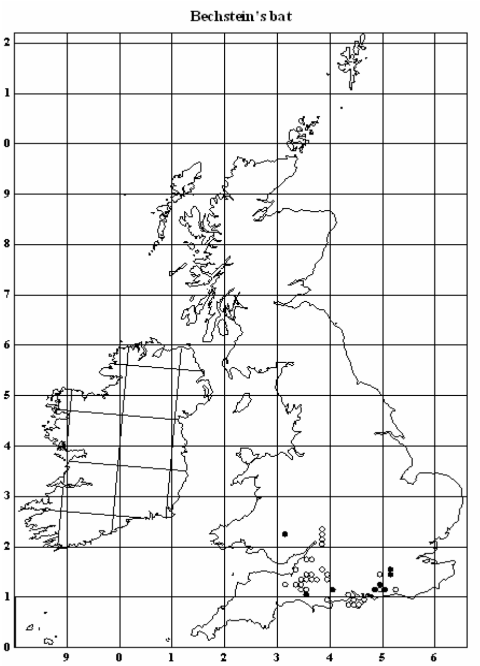
a) Leisler’s bat



b) Barbastelle bat



c) Bechstein’s bat



d) Grey long-eared bat

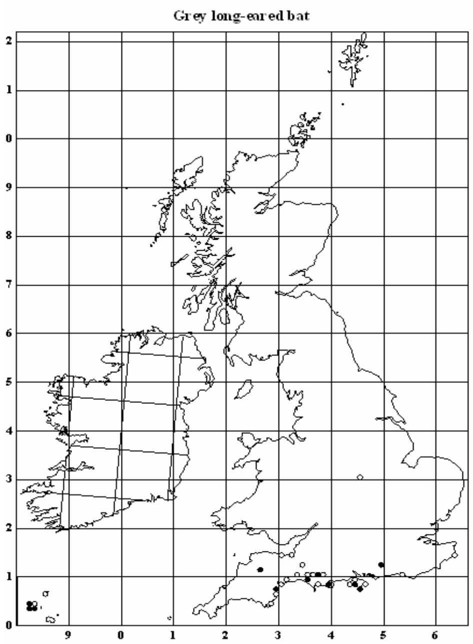


Figure 10.3 Distribution maps of four bat species frequenting woodland types W8, W10 and W16 (from Bat Conservation Trust [online], 2003).

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11. Soils and litter

Litter quality, organic matter dynamics and nutrient cycling in chestnut stands has been examined by a number of investigators. Ellenberg (1988) found that chestnut litter had a relatively short residence time (*c* 2 years) and a low C:N ratio, in which it compared favourably with oak, birch and beech (Figure 11.1). Litter breakdown in chestnut has also been extensively studied at Blean Woods in Kent by Anderson (1973a,b; 1975) in adjacent stands of stored chestnut and beech coppice, about 40-60 years of age. The study used litter bags of varying mesh size to differentiate between breakdown caused by soil fauna, microbial or abiotic agencies. In the chestnut plot, in which the soil had a predominantly mor-moder litter layer, chestnut litter was comminuted mainly by the abiotic processes of wind, rain (leaching), and hygroscopic movements caused by drying and wetting, resulting in leaf cracking along veins and midribs. Beech leaf litter showed less physical breakdown due to its smaller size and finer, more flexible veins and midribs, allowing the leaf to curl rather than crack on drying.

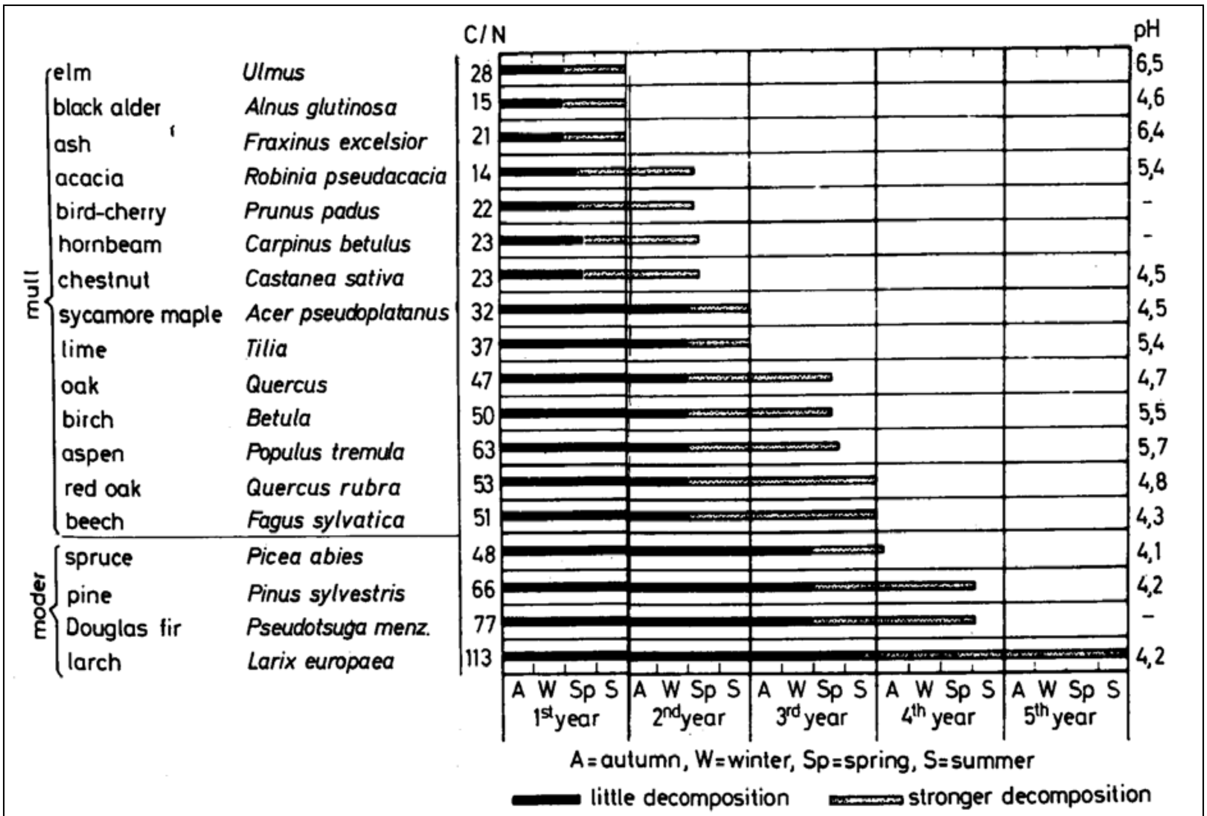


Figure 11.1 Average breakdown time of leaf litter of different species on a medium brown earth soil (from Ellenberg 1988, after Scheffer and Ulrich, showing pH and C:N ratio of fresh litter). Reproduced with kind permission from Cambridge University Press.

In the beech plot, where the litter layer was of a mull-moder type, a small population of earthworms was present and relative litter losses due to soil fauna were much greater here. Chestnut leaves were more readily removed than beech leaves from the coarse-mesh litter bags at this site. Over a period of 31 months, total percentage losses from the coarse mesh litter bags were greater for chestnut than beech (Table 11.1), nearly all the chestnut litter having disappeared within this period. However, Anderson also pointed out that losses from litter bags significantly underestimated the breakdown rates that would normally be expected in the field.

Table 11.1 Percentage losses of chestnut and beech leaf litter from coarse (7mm) and fine (175µm) mesh litter bags after 31 months in the field at Blean Woods, Kent (after Anderson 1973a)

Leaf litter type	**mor-moder site (chestnut coppice)**		**mull-moder site (beech coppice)**	
	coarse	**fine**	**coarse**	**fine**
beech	40	45	58	56
chestnut	77	64	92	66

Anderson (1973b) also showed that the soil fauna fed more on chestnut than beech, and that this difference was not directly related to the lower C:N ratios in chestnut litter. Initially the high polyphenol and tannin contents of the litter reduced feeding activity, and the soil fauna did not attack it for the first six months, when some of these compounds had been leached. However, chestnut leaf palatablity was still greater when it contained higher polyphenol concentrations than beech. The latter also contained more protocatechuic and gallic acids, thought to be inhibitory to soil fauna. After 18 months, the litter of the two species contained similar numbers of soil fauna and species (Anderson 1975).

Decomposition of litter from chestnut, *Quercus petraea, Q.ilex* and *Fagus sylvatica* stands growing in the Cevennes region of France has also been investigated by Cortez and his co-workers both in the laboratory and the field (Cortez and others 1996; Cortez 1998; Cortez and Bouché 1998; 2001). Under laboratory conditions, mineralisation of fresh leaf litter followed the sequence *Q.petraea* >*Q.ilex* =*Fagus* > *Castanea*, but for the previous year's litter the order was *Castanea* =*Q.petraea* >*Fagus* (as no *Q. ilex* litter was present for the previous year). Decomposition rates of the previous year's litter were linked to litter quality indicators such as lignin content, which in the case of *Castanea* was lower than in the other species. *Castanea* also had a relatively higher percentage of hydrosoluble compounds, such as tannins and polyphenols, which tend to be mineralized quickly.

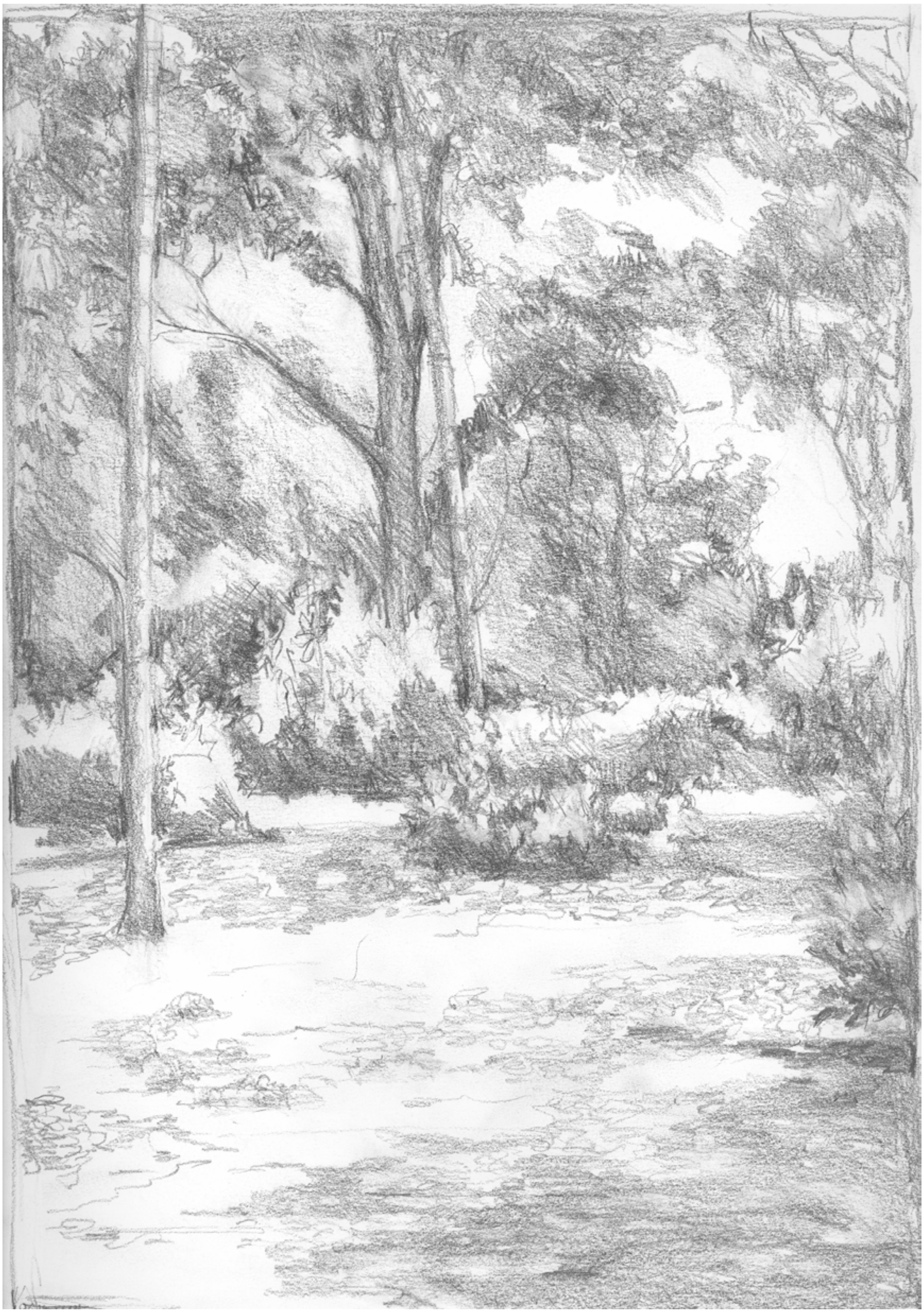
Differences in the palatability of the different leaf litters to earthworms were also demonstrated by Cortez (1998) and Cortez and others (2001). Using measurements of respired CO_2 under laboratory conditions, earthworm *Lumbricus meridionalis* biomass in both fresh and composted litters followed the general sequence *Castanea* > *Quercus petraea* > *Q. ilex* > *Fagus sylvatica*. Chestnut litter also decomposed faster at the field sites than the other species, reducing to 66-88% during the first two years in soils varying from pH 4.8-5.4 (Cortez 1998). Like Anderson, Cortez and others found that the litter underwent a preliminary microbial decomposition in the soil during the first year, after which it became more palatable to earthworms as the levels of aromatic phenolic compounds and primary and carboxylic alcohols decreased over time.

There has been some speculation that compounds in chestnut leaf litter may inhibit not only the soil fauna, but also plants and the microbial community. Litter of several members of the Fagaceae has been shown to contain allelopathic substances, such as coumarins and phenols, which inhibit the germination of herbs and grasses (Fisher 1980). In the case of the American chestnut *C. dentata*, Vandermast and others (2002) demonstrated that seedlings of eastern hemlock *Tsuga canadensis* and rosebay rhododendron *Rhododendron maximum*, species co-occurring within the range of chestnut in the Southern Appalachians, were strongly affected by the leaf litter leachate applied under laboratory conditions. From this evidence they suggested that allelopathic mechanisms could partly explain the dominance of chestnut in eastern deciduous forests prior to the arrival of chestnut blight *Cryphonectria parasitica*. Current forest communities could therefore be the result of the removal of this allelopathic influence, allowing species such as *Tsuga* to invade former chestnut habitat.

Little corresponding work has been carried out on *C. sativa* or its native tree associates in Britain, but Basile and others (2000), working in Italy, have demonstrated both allelopathic and antibacterial properties of chestnut leaf extracts. They found that fresh leaf extracts contained flavonoids, isoflavonoids and glycosides of luteolin and apigenin which had pronounced antibacterial effects against eight strains of Gram-positive and Gram-negative bacteria. The highest anti-bacterial activity was shown with quercetin, apienin, morin, naringin, galangin and kaempferol, of which quercetin, rutin and apigenin inhibited seed germination and root and epicotyl growth of radish *Raphanus sativus*.

These results suggest that further litter studies are worth pursuing to establish whether allelopathic mechanisms do exist in chestnut-dominated woodland, and whether these are shared with associated native trees, such as *Quercus* and *Betula*. Observations of dense litter accumulations under chestnut stands are not necessarily confirmatory evidence of allelopathy, but may simply indicate higher levels of stocking and productivity than in mixed coppice stands. The limited evidence does, however, suggest that chestnut litter is broken down at least as quickly as that of its native tree associates in Britain on similar sites, and that nutrient cycling, litter turnover rates and the soil fauna populations are not adversely affected.

Part III – Conservation strategies and management



12. Planning for biodiversity

12.1 Conservation policies: Biodiversity Action Plans

The nature conservation status of chestnut woodland is somewhat anomalous when stands are located on ancient sites containing remnants of native woodland. Indeed, such sites often enjoy the protection of National and local biodiversity action plans (BAPS, through Habitat Action Plans (HAPS) and Species Action Plans (SAPS), especially where the coppice habitat supports rare species dependent on young growth stages for their survival.

In 1984 the Government introduced a consultative paper on broadleaves policy that included a recommendation to return to coppice working which aimed, amongst other objectives, to mitigate losses of important species (Forestry Commission 1984). Current government policy on biodiversity relevant to lowland, broadleaved woodland is set out in the *Working with the grain of nature: a biodiversity strategy for England* (Department of Environment, Food and Rural affairs 2002). The UK Biodiversity Action Plan (Department of Environment 1994) continues to be taken forward under the UK Biodiversity Partnership and through policies outlined in *Sustainable Forestry: the UK programme* (Forestry Commission 1994), *The UK Forestry Standard* (Forestry Commission 1998) and the *England Forestry Strategy* (Forestry Commission 1999). Recommendations for sustainable forest management, drawing on these policies, are set out in the Forestry Commission's practice guidelines on the management of semi-natural woodlands (Forestry Commission 1994) and have been adopted into the UK Woodland Assurance Scheme (UKWAS Steering Group 2000).

12.1.1 The UK Biodiversity Action plan (1994)

According to the UK Biodiversity Action Plan (UKBAP; Department of Environment 1994), coppice woodlands promote early successional species, but they contain few ancient trees, which are important for specific lichens and invertebrates and not much deadwood, which is important for some fungi, mosses and invertebrates. On the other hand, the UKBAP also mentions the value of coppice-with-standards in maintaining populations of plants and animals that have been lost as coppicing declined during the last century.

12.1.2 Woodland types substituted by chestnut with reference to the woodland LBAPS and HAPS

The Habitat Action Plan most relevant to chestnut woods is the Lowland Mixed Deciduous Woodland HAP (JNCC 2003, draft version 5) as this is concerned with woodland types where most chestnut substitution has occurred, ie W8, W10 and W16 communities of the NVC (Section 3.3). This identifies lowland mixed deciduous woodland as mainly consisting of these three NVC types (Table 12.1)

In the 1980s the Nature Conservancy Council estimated the extent of this type of woodland at about 250,000 ha (Lowland Mixed Deciduous HAP, JNCC 2003). The somewhat smaller figure given by the *Forestry Commission Practice Guide* (Practice Guide 3, 1994) of 130 – 160,000 ha excludes some areas of lowland beech and yew and lowland acid beech and oak woods described in other *Practice Guides* (1 and 2, respectively). Work is under way to update these figures.

Table 12.1 Woodland community types covered by the Habitat Action Plan for Lowland Mixed Deciduous Woodland (JNCC 2003)

NVC code	Community description	Notes
W8	*Fraxinus excelsior – Acer campestre - Mercurialis perennis* woodland	Ancient or recent
W8a	*Primula vulgaris – Glechoma hederacea* sub-community	Ancient or recent
W8b	*Anemone nemorosa* sub-community	Ancient or recent
W8c	*Deschampsia cespitosa* sub-community	Ancient or recent
W8d	*Hedera helix* sub-community	Mostly secondary woodland
W10	*Quercus robur – Pteridium aquilinum – Rubus fruticosus* woodland	
W10a	Typical sub-community	
W10b	*Anemone nemorosa* sub-community	
W10c	*Hedera helix* sub-community	
W10d	*Holcus lanatus* sub-community	
W16	*Quercus* spp. – *Betula* spp. – *Deschampsia* flexuosa woodland	
W16a	*Quercus robur* sub-community	

The Lowland Mixed Deciduous Woodland HAP identifies lowland mixed deciduous woodland as one of the richest habitats for wildlife, stating that "in many eastern counties it forms the main reservoir of semi-natural habitat in the agricultural matrix". It includes the bluebell woods for which the UK is renowned and has particular responsibility, having over 25% of the world resource (Kent Biodiversity Action Plan Steering Group 1997). This type of ancient woodland has decreased in area by about 30-40% (*c* 90,000 ha) over the last 50 years (JNCC 2003).

Sweet chestnut occupies approximately 18,788 ha (National Woodland Inventory figure) of the 130–160,000 ha of this habitat type, or about 10-12% of all Lowland Mixed Deciduous Woodland.

12.1.3 Key species

Species Action Plans linked to the Lowland Mixed Deciduous Woodland HAP are currently being drafted. The UKBAP species for which this habitat is important include: butterflies, for example the heath fritillary *Mellicta athalia*, and pearl-bordered fritillary *Boloria euphrosyne* and various moths; birds such as the song thrush *Turdus philomelos* and mammals such as the dormouse *Muscardinus avellanarius*. Key species of Lowland Deciduous Woodland are also listed in the relevant county Biodiversity Action Plans. The latter also produce plans and actions for notable species where these are scarce or declining: examples are given for Kent, Sussex and Surrey (Table 12.2).

Information on the present or potential impact of sweet chestnut planting is available only for a small number of key BAP species (see Part II). Generally the impact of sweet chestnut on these species is either neutral or detrimental. There is little evidence to suggest that chestnut, however it is managed, contributes to a more favourable habitat for any key species. It may provide a further food source for species such as the waved carpet moth *Hydrelia sylvata* (see Section 8.2) or an additional habitat for some tooth fungi (see Section 7.2), but no BAP species appears to be entirely dependent on sweet chestnut, the tree or the habitat.

Table 12.2 Woodland species listed in the UK Biodiversity Action Plan and Local Plans for Kent (1), Surrey (2) and Sussex (3)

Common name	Latin name	1	2	3
UKBAP SPECIES				
Invertebrates – general				
A Mining Bee	*Andrena ferox*			
Southern wood ant	*Formica rufa*		x	
Shining guest ant	*Formicoxenus nitidulus*		x	
A chafer	*Gnorimus nobilis*		x	
A Cranefly	*Lipsothrix nervosa*		x	
Leaf rolling weevil	*Byctiscus populi*		x	
A Weevil	*Procas granulicollis*			
Weevil	*Procas granulicollis*		x	
Saproxylic beetles			x	
A cardinal click beetle Saproxylic beetle	*Ampedus nigerrimus*			
Saproxylic beetle	*Ampedus ruficeps*			
Saproxylic beetle	*Ampedus rufipennis*		x	
Saproxylic beetle	*Dryophthorus corticalis*			
Saproxylic beetle	*Elater ferrugineus*			
Saproxylic beetle	*Eucnemis capucina*			
Saproxylic beetle	*Gnorimus variabilis*			
Saproxylic beetle	*Lacon querceus*			
Saproxylic beetle	*Hypebaeus flavipes*			
Blue Ground Beetle	*Carabus intricatus*			
6 spotted pot beetle	*Cryptocephalus sexpunctatus*		x	
a Ground Beetle	*Dromius quadrisignatus*			
Bast bark beetle	*Ernoporus tiliae*		x	
Maple wood-boring beetle	*Gastrallus immarginatus*			
Violet Click Beetle	*Limoniscus violaceus*			
Stag beetle	*Lucanus cervus*		x	x
A click beetle	*Megapenthes lugens*			
Eyed longhorn beetle	*Oberea oculata*			
Butterflies				
High brown fritillary	***Argynnis adippe***	x		

Common name	Latin name	1	2	3
UKBAP SPECIES				
Pearl bordered fritillary	***Boloria euphrosyne***	x	x	
Heath fritillary	***Mellicta athalia***	x		
Moths				
New Forest Cicada	***Cicadetta montana***			
White-spotted Pinion	*Cosmia diffinis*		x	
Heart Moth	*Dicycla oo*			
Waved carpet	*Hydrelia sylvata*		x	
Orange Upperwing	*Jodia croceago*		x	
Drab looper	*Minoa murinata*		x	
Scarce Merveille du jour	***Moma alpium***	x		
Double Line	*Mythimna turca*			
Clay Fan-Foot	*Paracolax tristalis*		x	
Common Fan-foot	*Pechipogo strigilata*		x	
Argent and sable	***Rheumaptera hastata***		x	
White-line snout	*Schrankia taenialis*		x	
Olive crescent	*Trisateles emortualis*			
Square-spotted clay	*Xestia rhomboidea*		x	
Amphibians				
Great Crested Newt	*Triturus cristatus*			
Birds				
Nightjar	***Ccaprimulgus europaeus***	x	x	
Wryneck	*Jynx torquilla*			
Spotted flycatcher	*Muscicapa striata*		x	
Bullfinch	*Pyrrhula pyrrhula*		x	
Song thrush	*Turdus philomelos*		x	x
Mammals				
Barbastelle bat	*Barbastella barbastellus*		x	
Dormouse	*Muscardinus avellanarius*	x	x	
Bechstein`s Bat	*Myotis bechsteinii*		x	
Pipistrelle bat	*Pipistrellus pipistrellus*		x	x
Plants				
Veilwort	*Pallavicinia lyelli*		x	

Common name	Latin name	1	2	3
UKBAP SPECIES				
Mosses				
Spreading leaved beardless moss	*Weissia squarossa*		x	
Knothole Moss	*Zygodon forsteri*			
Liverworts and lichens				
Tree Catapyrenium	*Catapyrenium psoromoides*			
a Lichen	*Chaenotheca phaeocephala*			
New Forest beech-lichen	*Enterographa elaborata*			
a Lichen	*Enterographa sorediata*			
a Lichen	*Graphina pauciloculata*			
a Lichen	*Pseudocyphellaria norvegica*			
Fungi				
Sandy Stilt Puffball	*Battarraea phalloides*			
Royal bolete	*Boletus regius*		x	
Devil`s Bolete	*Boletus satanas*			
Oak polypore	*Buglossoprous pulvinus*		x	
Hedgehog fungus	***Hericium erinaceum***		x	
Tooth fungus	*Hydnellum aurantiacum*			
Tooth fungus	*Hydnellum ferrugineum*			
Tooth fungus	*Sarcodon scabrosus*			
Tooth fungus	*Hydnellum concrescens*			
Tooth fungus	*Hydnellum scrobiculatum*			
Tooth fungus	*Hydnellum spongiospies*			
Tooth fungus	*Phellodon confluens*			
Tooth fungus	*Phellodon tomentosus*			
Tooth fungus	*Phellodon melaleucus*			
Tooth fungus	*Sarcodon glaucopus*			
Ascomyte Fungus	*Hypocreopsis rhododendri*			
Earth-Tongue	*Microglossum olivaceum*			

Common name	Latin name	1	2	3
LOCAL HAP SPECIES				
Invertebrates - general				
A hoverfly	*Eumerus ornatus*		x	
Ash black slug	*Limax cinereoniger*		x	
A hoverfly	*Pocota personata*		x	
Black-headed cardinal	*Pyrochroa coccinea*			x
Butterflies				
Silver washed fritillary	*Argynnis paphia*	x	x	
Purple emperor	*Apatura iris*	x	x	
White admiral	*Ladoga camilla*	x		
Duke of Burgundy	*Hamearis lucina*	x		
Wood white	*Leptidea sinapsis*	x	x	
Small pearl bordered fritillary	*Boloria selene*	x	x	
Brown hairstreak	*Thecia betulae*		x	
Moths				
Triangle	*Heterogenea asella*	x		
Plume prominent		x		
Sub-angled wave	*Scopula nigropunctata*	x		
Lesser belle moth	*Colobochyla salicalis*	x		
Clifden non pareil moth	*Catocala fraxini*	x		
Festoon	*Apodia limacodes*		x	
Mocha	*Cyclophora annulata*		x	
Cloaked carpet	*Euphyria biangulata*		x	
Water carpet	*Lampropteryx suffumata*		x	
Beautiful carpet	*Mesoleuca albicillata*		x	
Pauper pug	*Eupithecia egenaria*		x	
Broad-bordered bee hawk	*Hemaris fuciformis*		x	
Orange footman	*Eilema sorocula*		x	
Small black arches	*Meganola stricula*		x	
Star-wort	*Cuculia asteris*		x	
Pale eggar	*Trichiura crateagi*		x	
Birds				
Hawfinch	*Coccothraustes coccothraustes*	x	x	
Wood warbler	*Phylloscopus sibilatrix*	x	x	
Redstart	*Phoenicurus phoenicurus*	x	x	

Common name	Latin name	1	2	3
LOCAL HAP SPECIES				
Firecrest	*Regulus ignicapilus*	x	x	
Hobby	*Falco subbuteo*	x		
Goshawk	*Accipter gentilis*	x	x	
Crossbill	*Laxia curvirostra*	x	x	
Nightingale	*Luscinia megarhynchos*	x	x	
Lesser spotted woodpecker	*Dendrocopus minor*	x		
Honey buzzard	*Pernis apivorus*		x	
Buzzard	*Buteo buteo*		x	
Mammals				
Yellow-necked mouse	*Apodermis flavicollis*		x	
Serotine bat	*Eptesicus serotinus*		x	
Brandt's bat	*Myotis brandtii*		x	
Whiskered bat	*Myotis mystacinus*		x	
Natterer's bat	*Myotis nattereri*		x	
Noctule bat	*Nyctalus noctula*		x	
Plants				
Wood anemone	*Anemone nemorosa*	x		
Hay scented buckler fern	*Dryopteris aemula*	x		
Helleborines		x		
Lady orchid	*Orchis purpurea*	x		
Fly orchid	*Ophrys insectifera*	x		
Birds nest orchid	*Neottia nidus-avis*	x		
Small-leaved lime	*Tilia cordata*	x		
Butcher's broom	*Ruscus aculeatus*	x		
Box	*Buxus sempervirens*	x	x	
Bluebell	*Hyacinthoides non scripta*	x		
Tunbridge filmy fern	*Hymenophyllum tunbrigense*	x		
Starved wood sedge	*Carex depauperata*		x	
Green hound's toungue	*Cynoglossum germanicum*		x	
Herb Paris	*Paris quadrifolia*		x	
Common Solomon's seal	*Polygonatum multiflorum*		x	
Wild service tree	*Sorbus torminalis*		x	
Marsh fern	*Thelypteris palustris*		x	
Mistletoe	*Viscum album*		x	

Common name	Latin name	1	2	3
LOCAL HAP SPECIES				
Lichens				
A lichen	*Fellhanera bouteillei*		x	
Moss				
A moss	*Seligera paucifolia*		x	

12.2 Action plan targets

12.2.1 Management issues

Cessation of traditional silvicultural practices has led to a reduction in structural diversity within the woods, in particular the loss of open space. The Lowland Mixed Deciduous Woodland HAP recommends the encouragement of a blend of management regimes, including minimum intervention, coppicing and high forest management within regions and across the habitat type. In the Forestry Commission Practice Guides, continued coppicing is recommended where:

- the coppice is still being cut;
- previous coppicing occurred within the past 50 years;
- wildlife species which prosper in coppice (such as dormice and nightingales) are present;
- deer populations are low, and
- traditional markets still exist.

Maintaining coppice cycles in small woods is emphasised to retain small patches of habitat for sensitive species that would otherwise be lost, and coppice-with-standards is recommended to increase habitat diversity, keeping standard trees between 40-50% of the canopy cover.

Where coppicing has long ceased, the default position is high forest management or minimum intervention. The former was considered the most suitable for larger woods in the *Forestry Commission Practice Guides*, both for Lowland Acid Beech and Oak and Lowland Beech and Ash Woods, although retaining patches of coppice was thought desirable. Coppice conversion to uneven-aged silvicultural systems is recommended in smaller woods in order to maintain a range of age-classes. It is questionable, however, that this would be effective in very small woods where patches of each age cohort would occupy less than the 'minimum dynamic' area required by some species.

12.2.2 Restoration guidelines

The Lowland Mixed Deciduous Woodland Habitat Action Plan includes a recommendation to restore ancient replanted woodland sites that have been substantially replanted with conifers in the last 50 years, or are dominated by non-native tree species. The overall target is to convert 25,000 ha of replanted ancient woodland to native broadleaves by 2025, or about 10% of the existing resource. Interim targets are to initiate the restoration of 7,000 ha by 2004 and a further 18,000 ha by 2015. Some of this might in small part be achieved by converting chestnut monocultures, especially where these are comparatively recent, nineteenth century plantations.

Significantly, there is no explicit recommendation in the Habitat Action Plan as to whether to treat chestnut as an exotic. In the *Forestry Commission's Practice Guide 3* on Lowland Mixed Broadleaved woodland, it is stated that '*… chestnut and beech may be retained as part of the mixture on the ground, ie their spread should not be extended by planting*'. Chestnut is not mentioned at all in the other relevant Practice Guides (1 and 2) or in the

Forestry Commission's recent guide on the *Restoration of Native Woodland* (Thompson and others 2003). Reversion of chestnut to semi-natural ancient woodland will probably, therefore, remain a very low priority after conifer removal. Forced reversion would also carry a risk that the removal of chestnut stools could be more detrimental to some species of high conservation value (which may already be utilising the chestnut habitat) than maintaining regular rotations *in situ*.

The lack of definite guidelines in relation to the restoration of chestnut woodlands is endorsed by Rackham (1980), Peterken (2000) and Spencer (2002). None of the authorities appear to regard sweet chestnut as particularly invasive or a serious threat to the established ecology of ancient woodland sites, such that there is no urgency to pursue restoration policies. Generally, this review confirms these conclusions.

12.2.3 Woodland expansion guidelines

The Lowland Mixed Deciduous Woodland Habitat Action Plan includes a commitment to expand the area of lowland mixed deciduous woodland, preferably through natural colonisation, but also by planting site-native species or species of local genetic provenance. The specific target is to establish 25,000 ha on unwooded sites or in recent conifer plantations by 2015, initiating 50% of this establishment by 2010. As it is not site-native, this appears to exclude chestnut as a candidate for woodland expansion, although it may be acceptable in mixed plantings with other (native) broadleaves. *Castanea* is also excluded from Forestry Commission design prescriptions given for creating new native woodlands (Rodwell and Patterson 1994).

In terms of nature conservation objectives, excluding non-native sweet chestnut from new plantings seems an appropriate policy. As an economic crop the arguments for new plantings of sweet chestnut are weak; coppice is already insufficiently managed due to a lack of markets (Section 5.2) and its adoption as a timber crop by growers seems unlikely at present.

12.3 Local Woodland Habitat Action Plans

The Local Plans of particular relevance to this review are those of Kent, Sussex and Surrey, as an estimated 58% (Forestry Commission 2000) of chestnut woodland occurs within these south-eastern counties.

12.3.1 Management issues

There are a number of issues identified by the Kent, Surrey and Sussex Woodland Plans that directly affect sweet chestnut. Issues relating to coppice and sweet chestnut include:

- Unmanaged and neglected coppice.
- Loss of rides, glades and patches of heath.
- Reduction in the area of coppice woodland.
- Increasing areas of coppice reverting to high forest.
- Low wildlife/biodiversity value of sweet chestnut coppice.
- Damaging or potentially damaging animals, eg deer.
- Lack of markets for coppice produce and resulting financial constraints.

- Lack of interest, expertise and incentives resulting in some unmanaged and unsympathetically managed woodland.
- Need for more incentives to encourage or support positive management.
- Potential effects of climate change.

In Kent, somewhere in the region of 60% of chestnut is currently unmanaged (Kent Biodiversity Action Plan Steering Group 1997). Sussex and Surrey are similarly affected. The decline of species such as dormice or the wood white butterfly *Leptidea sinapis* resulting from the cessation of coppicing and the overgrowth of rides is general, occurring in all stand types as well as chestnut.

12.3.2 Aims and actions for positive management

The major aims and proposed actions from the LBAPs relevant to coppice and sweet chestnut are as follows:

- Increase area of managed coppice/reinstate coppice management.
- Increase biodiversity value of sweet chestnut plantations.
- Consider other methods of enhancing biodiversity where coppice management cannot be reinstated.
- Control species that threaten woodland with high biodiversity value, eg deer, squirrels and rabbits.
- Promote positive management of ancient semi-natural woodland and replanting with or regeneration of native species.
- Restore ancient replanted woods to semi-natural conditions.
- Review financial incentives and wood markets.

Increasing the area of managed coppice is one of the main aims of the Local Plans. The Kent Plan (currently under review) proposes an increase in coppice management from 40% to 50% by 2007 with the aim that in 50 years time 75% of the coppice will be managed. Chestnut coppice is not specifically mentioned in these target figures and the proportions of different woodland types to which this prescription applies is also uncertain. Such targets also seem unrealistic in view of the evidence for coppicing density in the county (Section 5). The Kent Plan suggests that priority should be given to reinstating coppicing on ancient and also designated conservation sites, eg SSSI and SNCI woodlands.

In contrast, the Surrey and Sussex Plans do not give specific restoration targets, while the latter even suggests that reinstating coppicing may not always be appropriate. This is because sites neglected for a long time may no longer retain the wildlife dependent on the coppice cycle and may not easily regain these species. The Sussex Plan recommends that such sites are converted to high forest, with a coppice or shrub understorey rather than a coppice-with-standards system. None of these recommendations refer specifically to chestnut.

Increasing the structural diversity of sweet chestnut plantations is addressed by the local Plans as another key objective. The Kent Plan suggests retaining standard trees and allowing some coppice to develop into high forest, while in Surrey the Plan advocates practices such as ride management, coppicing, thinning and selective felling. All plans recommend monitoring key species. Traditional coppice-with-standards is considered the preferred system for biodiversity, while the Surrey Plan also advises retaining remnants of 'old growth' woodland within coppices, and new planting to encourage interconnectedness.

Policy

Poor financial sustainability of coppicing is identified as one of the main reasons for its neglect. All Local Plans suggest reviewing financial incentive measures, such as the Woodland Grant Scheme. For example, from 1996-1998 the Woodland Grant Scheme provided Challenge funds, under the Woodland Improvement Grant, specifically to support coppicing for restricted butterfly species in areas of East Kent and the High Weald in undermanaged woods.

The importance of developing wood markets is also highlighted in several Plans. Encouraging marketing organisations such as the Weald Woodnet, the Woodland Enterprise Centre and the Wood Products Producer Group are examples. The aim of the latter is *to address decline in woodland owners' income, to promote the traditional coppice management system, to enhance biodiversity, landscape and the recreational value of woodlands and maintain local jobs within the woodland industry. The project will set up a Producers' Group dealing with wood products. Training will enable the members of the group to formulate management plans and get certification for their woodlands, therefore adding value to local wood products*. (EDU, Kent County Council 2003).

A key wood products marketing tool is the Forest Certification Scheme. The Forest Stewardship Council (FSC) label guarantees forest products are from environmentally sustainable sources. This 'green-labelling' has proved very beneficial for marketing; the demand for sustainable products currently exceeds supply (Goodall 2002). The UK Woodland Assurance Scheme (UKWAS) is endorsed by the FSC and products from UKWAS forests can also display the FSC label. Although there are obvious benefits, certification is likely to be slow to be adopted by small woodland owners who find the cost, bureaucracy and administration of the scheme onerous. Take-up has been slow and under 20% of Kent's woodlands have achieved certification to date.

13. Discussion and conclusions

The literature consulted during this review suggests that few, if any species are uniquely dependent on chestnut in its range in southern Britain. Most species using chestnut directly as a host were also present on other Fagaceae, or other trees or shrubs in the same environment. The number and variety of different taxa associated with chestnut also appears to be lower than that of native species. A summary of the ecological effects of chestnut on different species groups is given in Table 13.1.

In common with other plantation crops, the uniformity of chestnut monocultures and the brevity of the coppice cycle inevitably reduces the number of available niches for wildlife. However, for specialists requiring young forest growth stages, the crop is a useful surrogate for native broadleaves and is benign in the sense that it diversifies the woodland habitat without essentially changing its physiognomy, unlike evergreen and conifer species introductions (Peterken 2001). The system of relatively small but varying coupe sizes, together with rides and open spaces in worked chestnut coppice, also adds to diversity at both a compartment and whole forest scale. Furthermore, there is no strong evidence that the species is particularly invasive in Britain or is threatens the habitat of ancient woodland communities dominated by oak and birch.

To increase species diversity in chestnut stands, two basic guidelines emerge from the literature:

1. **Diversify the age structure**. This can be achieved relatively quickly by increasing the rotation length, allowing stands to emerge from the closed-canopy stage and develop into high forest. Singling of coppice stools may further assist with this process, but abandonment would also have a similar effect in the long run, leading to self-thinning and eventually the diversification of the canopy through natural disturbance. Variable-length rotations (ie some long, some short) are another possibility, allowing high forest elements or extended-rotation coppice to alternate with normal coppice, but will depend on wood markets and managers' perceptions of the susceptibility of promoted coppice stems to ring-shake or windthrow. Within compartments, standard trees were found consistently to improve species diversity in a number of species groups. Standards can be introduced, or coppice stems (including chestnut) or natural regeneration promoted.

2. **Diversify the species structure.** As mixed broadleaved coppice stands appear to be generally richer in species, some dilution of chestnut monocultures may be thought desirable. At a compartment level this could be achieved by a) stool grubbing or poisoning, substituting native species, b) selective cutting and thinning to promote natural regeneration and c) abandonment, allowing stands to diversify naturally. Alternatively in uneconomic crops these treatments can be applied at a whole compartment level, producing a greater diversity at the whole forest scale.

Chestnut silviculture depends not only on available markets, but also on current nature conservation interests and prejudices, and the cultural acceptance of the species in Britain. These in turn will be strongly influenced by future events, both economic and natural, including the viability of timber markets, the introduction of disease, and the long-term influence of global warming.

13.1 Reversion and abandonment in chestnut stands

The evidence of progressive abandonment of chestnut and other coppice crops over the past 50 years, following market failure, is compelling. Although some large estates in southern England are still able to maintain pure chestnut coppice working, it is foreseeable that smaller, less viable parcels will suffer increasing neglect. No long-term studies have been carried out into the question of how resilient (*sensu* Connell and Slayter 1977) chestnut plantations might be to natural invasion, or whether in the long term they will be able to maintain and reproduce themselves. However, one study has investigated non-native holm oak coppice *Quercus ilex* in the Cévennes region of southern France (Bacilieri and others 1994). Experimental sowing of seeds of the downy oak *Quercus pubescens*, native to the region, showed consistently better germination and establishment compared with similar sowings of *Quercus ilex* across a range of coppice densities, suggesting that the native *Q. pubescens* could again become dominant in the event of abandonment. Chestnut seeds introduced under chestnut orchards and coppices in the same region also did well within their optimum altitudinal range, particularly in less dense stands, but unfortunately these authors carried out no parallel sowings of *Quercus pubescens* within the chestnut crop.

On ancient woodland sites already converted to chestnut, native trees and shrubs present in the canopy or in the surrounding landscape may be expected to gradually invade and diversify these introduced stands. In Lady Park Wood, Gwent, a mixed-species coppice-with-standards crop that was felled in 1940-44 and naturally regenerated, followed by self-thinning, developed areas with a marked diversity of stand structure and species composition after 40 years (Peterken and Jones 1987,1989). Although in the short term the coppice stems will dominate the canopy as even-aged high forest, at some point canopy gaps created by natural disturbances will provide regeneration niches for other species. Historical evidence of naturalized chestnut stands occupying ancient woods (Section 2), together with the recognized ability of the species to regenerate spontaneously, suggests that it is capable of maintaining a share of the canopy across a wide range of woodland sites. Ultimately, mixed and uneven-aged stands will tend to replace monocultures.

13.2 The risk of disease and pests

A future cause of reversion, diversification and abandonment in chestnut stands would be the introduction of chestnut blight into the UK. Recently blight has begun to have more impact in northern Europe, where natural hypovirulence is still poorly established (Robin and Heiniger 2001). Canopy gaps created by diseased trees can be expected to have a major effect, especially if this in turn leads to the abandonment of affected stands. In a study of plant species diversity in chestnut stands in the Cévennes region of France, Gondard and others (2001) demonstrated that abandoned, blight-damaged groves were colonized rapidly by invading shrubs. The extent to which UK stands might be affected is difficult to predict, but in a study area in northern Portugal the disease had affected up to 10% of all trees in chestnut groves within 12 years of *Cryphonectria* having been confirmed in 1989 (Gouveia and others 2001). A detailed review of data of the mortality rates, susceptibility and recovery of trees in blight-affected regions of other European countries would provide a model for what might occur in the UK.

The risk of transfer of new pathogens into Britain may be significantly increased by climate change. Ink disease in chestnut is caused by two pathogens, *Phytopthora cinnamomi* and *P. cambivora.* Models of global warming of 3°C indicate that *P. cambivora* could extend its range northwards increasing its current activity in Britain (Figure 13.1, Lonsdale and Gibbs (2001). Not only may some diseases become more prevalent, but insect pests of chestnut such as *Curculio elephas* and *Cydia splendens* could become more widespread.

13.3 Effects of climate change

During the remainder of the 21st century, climate warming in southern Britain will have a major impact on forest ecosystems, resulting in the gradual reorganisation of their species composition over time. Future tree distributions can be modelled using bioclimatic data. These studies indicate that beech, for example, will contract northwards from its current range in southern Britain (Sykes and others 1996), while at the same time semi-natural woodland communities such as W8 and W10 may expand, the increased temperatures in the latter case favouring oak regeneration over birch (Ray and others 2002). Medium-high prediction scenarios of climate change show that accumulated temperatures and moisture deficits during the growing season are expected to increase by 50% and 15%, respectively, in the south and east of England by 2080.

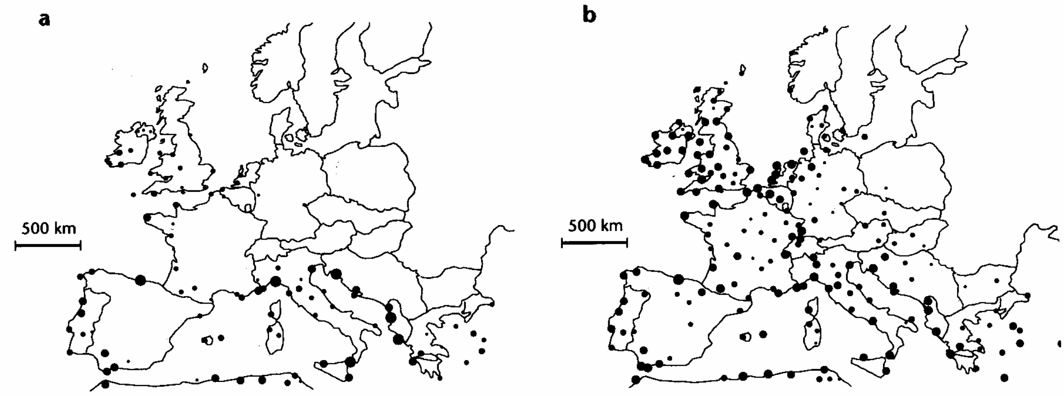


Figure 13.1 Shows a) current range of *Phytopthora cinnamomi* and b) predicted ranges modelled for an increase in temperature of 3°C (after Brasier and Scott).

(Dot size indicates relative activity of the fungus).

Enhanced temperatures and moisture deficits are likely to benefit chestnut, as similar conditions already apply over parts of its current naturalized range in southern Europe. In order to model future changes in forest composition, it is necessary not only to consider the optimum bioclimatic range of a species but also the dynamics of its growth and its interaction with other species. Sophisticated forest gap models that take account of the changing climate are still, however, subject to considerable margins of error inherent in climatic scenarios generated by General Circulation Models. A cautionary example of the sensitivity of inbuilt assumptions in forest ecosystem modelling is illustrated by Fischlin and others (1995), who explored the effects of climate change on forest composition in a gradient of three test sites in the Alps. At their highest elevation site, Bever in Swizerland (1708 m above sea level), forests are currently dominated by *Larix decidua, Pinus cembra* and some *Pinus mugo*. Assuming a temperature rise of 3.3°C (compared with pre-industrial levels) by 2070, the reference model predicted the disappearance of conifers and their replacement by a broadleaved forest consisting predominantly of Norway maple, sycamore and hornbeam. If, however, the actual rise were 1°C higher than that estimated by General Circulation models, up to 50% of the forest could then support chestnut (Figure 13.2).

Despite the uncertainty of global warming predictions, there are strong indications that, with the retreat of beech and birch, the potential range of chestnut could expand further in southern England and beyond. As more former coppice crops develop into high forest and the weight and frequency of masting increases with climate warming, chestnut may become an increasingly invasive and ‘aggressive’ component of ancient woods. At the same time there are a number of counterbalancing factors that could keep chestnut expansion in check. These include future epidemics of *Cryphonectria* and *Phytophthora*, increased predation of seed and seedlings by insects, small mammals and deer, and defensive nature conservation practices to remove chestnut. The number of variables and ecological linkages make predictions hazardous.

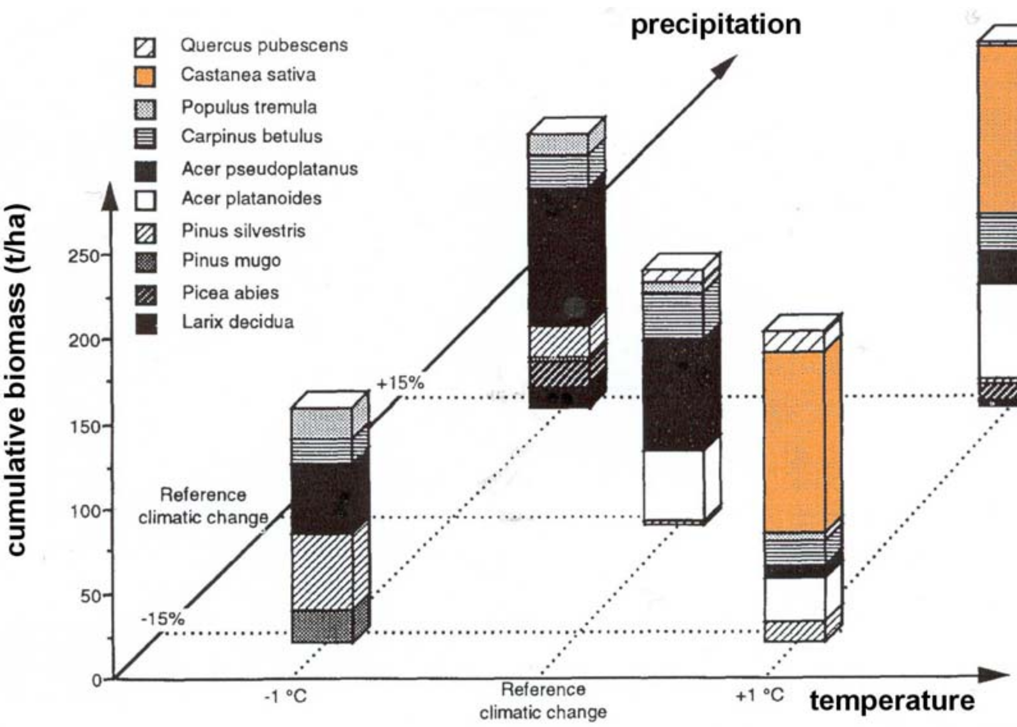


Figure 13.2 Predicted effects of climate change on forest composition at Bever, Switzerland for the reference climatic change senario (centre) and four alternative deviations from this model in relation to temperature and rainfall (from Fischlin and others 1995). Reproduced with kind permission from Elsevier.

13.4 Conservation objectives

Nature conservation strategies need to distinguish between the naturalized and artificial elements of the chestnut habitat. Guidance in Biodiversity and Habitat Action Plans is distinctly muted on the issue of chestnut restoration, given the existence of more pressing priorities, such as the removal of conifers from ancient woodland sites. However, in the case of historically-authenticated, ancient woods where chestnut has long been naturalized, such as Norsey Wood in Essex or Ellenden Wood in Kent, these stands have a claim to be recognised in conservation designations and protected accordingly. The situation is different for late-nineteenth century and recent coppice plantations which have no track record of long-term chestnut occupation and lack the evidence of ancient stools. In these situations the case for conservation is much weaker and their restoration or reversion to native woodland communities may be considered more appropriate. Naturally regenerated stands can be treated in a similar way, depending on the site's history.

Where there are specialist species such as migrant birds, nightjars, dormice and fritillary butterflies associated with young-growth chestnut stands, the case for maintaining a regular coppice cycle is clearly a high priority. Many such sites have already been designated for their nature conservation value, linked in turn to the presence of these species. As it cannot be assumed that other, undesignated chestnut coppice areas will continue to be worked on normal rotations, it is important to establish their status regarding specialist species in order to prioritise cases for conservation coppicing. Although Woodland Improvement Grants (WIGs) and their associated Challenge Funds have been concerned to maintain the coppicing cycle for particular wildlife species and to revive rural economies, chestnut coppice areas are not usually the primary target. WIG schemes have not been effective in preventing some areas of coppice from reverting to high forest, suggesting not only that the incentives are insufficient, but that local Biodiversity Action plans to increase the active area of coppicing are unrealistic.

Where there are no claims of species of conservation importance, several techniques are available to diversify and increase the wildlife interest of chestnut stands at the stand scale:

- introduce or increase standard tree densities up to 25-50 ha^{-1} to vary the stand structure, including promoting standards of chestnut;
- practise part-restoration, ie negative selection during thinning or singling of chestnut stands to high forest, reducing the dominance of chestnut to set target levels, eg 50% of the stand basal area;
- allow stands eventually to revert to high forest and self-thin, using minimum intervention;
- retain old, veteran trees.

At the compartment level:

- maintain a diversity of woodland communities and species associated with chestnut, designating some management units as ‘chestnut free zones’ to be replanted or naturally regenerated with native species;
- vary the coppice coupe size, with some larger areas of 0.5-1 ha to accommodate territories of summer migrants and to encourage small mammals;
- maintain open ride areas, especially in relation to specialist species requiring open conditions (Ferris 2000). Rides can also be linked to public access as part of the justification for restoration schemes;
- operate different-aged or multiple rotations within the chestnut crop to vary the structure over the forest area as a whole;
- revert to a continuous cover or small-group felling regime in less economically viable parcels.

Alternative management solutions for restoring or diversifying chestnut stands, together with the likely consequences of management, are given in Table 13.2.

13.5 Research questions

As might have been anticipated at the start, this review has raised more questions about the ecological impact of chestnut than it has succeeded in answering. However, there are a number of areas that would benefit from further study and research, including the following:

1. Surprisingly little appears to be known or recorded about the regeneration niche of chestnut and there are conflicting accounts on the frequency of spontaneous seedling establishment in the literature. Work is needed on the age of seed production in Britain, the precosity of flowering in coppice stands, flowering periodicity, quantities

of and seasonal variation in seed production, predation rates, agents of dispersal and conditions for seedling survival and recruitment.

2. Formal, replicated experiments are needed to validate the efficacy of stand restoration techniques on wildlife biodiversity, and to provide demonstration models. Basic treatments could be implemented at (preferably) a compartment or sub-compartment scale, including a) high forest reversion, b) altering standard densities, c) diversifying the chestnut underwood and d) normal coppicing 'controls'. Treated stands should be retained for long-term monitoring of the main taxonomic groups.

3. Resilience of chestnut stands to invasion could be examined by introducing native species into pure chestnut stands as seed or transplants, and *vice versa* by monitoring invasion through natural regeneration or planting of chestnut in semi-natural, ancient woodland (on an experimental scale). The possibility of alternation of regeneration of chestnut with other species, for example oak, should be examined;

4. Scenarios and models to predict possible future effects of chestnut blight, ink disease and other pathogens and pests are needed for the UK. Epidemiological patterns, drawing on European examples, should be investigated, as well as modelling using bioclimatic data.

5. The effects of global warming on the potential dominance and spread of chestnut needs to be examined experimentally and theoretically, taking account of genetic variation within British populations.

6. Market limitations for chestnut products need to be identified, in particular the scale of financial incentives needed to restore conservation coppicing. In the case of stored chestnut coppice, perceptions of, as well as the actual incidence of, shake defect as a barrier to timber markets needs further examination.

7. Some previous investigations have suggested that chestnut leaf litter may contain allelopathic substances that influence field layer species composition and may damage seedling recruitment. Further litter breakdown experiments are needed on a range of soil types, and any inhibitory effects on woody seedlings firmly established.

8. The economics of coppicing in different stand types and their impact on the chestnut industry needs further research. This would include an analysis of the costs of cutting chestnut coppice mixed with other species as well as in pure stands, the effects of summer cutting, and cutting for different products requiring varying rotation lengths.

Table 13.1 Summary of ecological effects of sweet chestnut planting on different species groups

Species group	Positive effects	Negative effects	Research questions
VEGETATION			
Stands often derive from semi-natural woodland communities and are therefore potentially species-rich compared with secondary woods. However, much chestnut planting has been on relatively infertile sites that are less species-rich than some other woodland communities. Stand-types in which chestnut does occur are relatively common in southern England.	Regular coppicing and associated disturbance opens up canopy, allowing the ground flora to develop and ruderal species to dominate temporarily, creating a wider species range. Specialist coppice species developing from buried seed, eg *Viola spp.* and *Melampyrum pratense*, may be important food plants for rare Lepidoptera	Rapid shading out of the ground flora restricts both plant and insect species diversity, thus reducing populations of foraging birds. Vernal species and shade-tolerant species are relatively unaffected. Young coppice stems support low numbers of epiphytic lichens.	Comparisons of managed and stored chestnut coppice are needed, preferably on similar soil types on adjacent sites, to fully evaluate the impact of chestnut grown as high forest on vegetation, including epiphytic lichens. Ideally 'controls' of native woodland communities would be included.
FUNGI			
In terms of numbers of species, the mycoflora is a major component of woodland biodiversity and is under-represented in Biodiversity Action Plan schedules. Recording is patchy and uneven across Britain. A variety of woodland structure, including over mature stands, will benefit this group.	Chestnut is second only to beech in hosting six UKBAP species. It supports four tooth fungi species not recorded on oak, but in the absence of beech is host to some of the rarer fungi. The endangered *Sarcodon scabrosus* appears to be particularly successful on chestnut. Fast-growing chestnut is likely to provide a substrate for decay fungi sooner than oak.	Lack of standing or fallen deadwood in regularly coppiced plantations reduces the potential for saprophytic species. Fewer species of fungi are found in chestnut than many of its tree and shrub associates in W10/W16 communities: none to date have been shown to be unique to chestnut. There is a strong risk that the parasitic fungus *Cryphonectria* could affect stands in the UK.	An under-researched group in woodland ecology. The co-hosting of rare species (eg tooth fungi) by chestnut and its native woody associates needs further research. Colonisation by saprophytic fungi of dead wood and comparative decay rates in chestnut and native woody species needs to be determined experimentally.
INVERTEBRATES			
Many invertebrate groups are under-recorded, making evaluations of biodiversity in different forest types difficult. Coppicing provides some advantages for certain species, but a range of stand types and rotations is likely to produce a greater variety of associated invertebrates. The number and variety of invertebrates associated with chestnut appears to be increasing as	As the relative abundance of chestnut increases, it can be hypothesised that more incidences of host adaptation will occur over time. The list of invertebrates associated with chestnut is growing, especially polyphagus insects dependent on its associates in native woodland. Chestnut is host to notable lepidopteran species, including *Hydrelia sylvata*, while active coppicing provides suitable	Studies suggest that lower numbers of invertebrates are dependent on chestnut compared with its native associates. Coppicing reduces structural diversity in woodlands and thus the potential number of niches, eg for insects dependent on flower and fruit production or beetles attacking decaying timber.	Further systematic surveys of invertebrates on chestnut stands adjacent to, or isolated from native woodland species would help to confirm patterns of polyphagus insect feeding in each woodland type. There have been relatively few studies of stand structure on species diversity in chestnut, or of species abundance in young or old stands.

Species group	Positive effects	Negative effects	Research questions
its range expands in the UK.	habitat for ground flora supporting *Mellicta athalia* and *Bolaria euphrosyne*.		
BIRDS			
Effects of coppice age on bird communities have been well studied in stand chronosequences at a number of sites. The widely ranging habitats and large territory sizes of some species make it difficult to determine the precise influences of pure chestnut crops compared with other stand types.	Species richness in coppice crops is initially high, prior to canopy closure, benefiting open-ground and migrant species, including notables such as nightjar and nightingale in southern England. The young growth structure provided by coppice crops provides good feeding and nesting habitat for these species.	Invertebrate biomass in pure chestnut stands appears to be lower than in native woodland, hence reducing foraging of summer migrants. Bird diversity is restricted in pure coppice stands, with hole-nesters and trunk and branch feeders restricted by a lack of mature trees.	Woodland structural diversity appears to be key, inferred from multivariate studies rather than by direct experiment. Experimental manipulations, such as diversifying chestnut stands by altering standard tree densities, reducing the proportion of chestnut and lengthening the coppice cycle, would provide more information.
MAMMALS			
Mammals are less well studied than birds, and there are fewer chronosequence studies or direct comparisons of different woodland types and structure in relation to mammal species diversity. Studies of browsing and palatability are highly site-specific.	Young coppice is suitable habitat for a wide range of small mammals before canopy closure, especially non-arboreal species. If standard trees are present or the coppice is overmature, reliable seed production may be a significant food source for small mammals (and wild boar) in otherwise poor mast years. Coppice structure and the edges of small compartments will tend to favour a number of bat species.	Chestnut coppice provides a less suitable habitat for dormice than mixed stands and rapid closure of the canopy eliminates ground layer vegetation, excluding many small mammals. Longer rotations than are commercially practiced are beneficial to dormice, but rotation compatibility with other notable species, such as fritillary butterflies and summer migrants, is a problem. Chestnut is palatable to deer which damage young coppice regrowth and understorey vegetation.	As with birds and invertebrates, structural diversity is important to small mammal populations, with standard trees and a wide age structure increasing the number of available niches. Deer browsing studies are inconclusive with regard to chestnut palatability and requires further study. The use of the coppice woodland habitat by bats is under-researched, particularly of rare species.
SOILS AND LITTER			
Decomposition and mineralisation studies are limited and do not cover the full range of soil types on which chestnut is grown. Comparative *in situ* and *ex situ* studies of litter types are needed.	Breakdown of chestnut litter appears to be more rapid than associated tree species, and its palatability to soil macrofauna, particularly earthworms, appears to be high.	Litter may have antibacterial and allelopathic properties, preventing seedling germination. Deep accumulations of litter under chestnut may be a reflection of rapid growth and high stocking densities.	Investigations of allelopathic properties of chestnut litter on ground vegetation and microbial biodiversity are needed. The effects of chestnut litter on nutrient cycling also need further study.

Table 13.2 Alternative management solutions for restoring or diversifying chestnut stands, with likely consequences of management

Action	Consequences of management
Maintain the active coppicing regime	Maintains the species requiring young growth stages but eliminates species requiring mature and late growth stages. Promotes populations of rare (BAP) species requiring open conditions, but is only worthwhile if they are already present on site or within dispersal range. Natural diversification of regularly coppiced stands will be slow due to the rapid re-growth of chestnut after cutting and slow rates of stool mortality.
Intervene to reduce chestnut dominance	Increases the proportion of other site-native trees and shrubs, which may need to be introduced if natural regeneration sources are too far distant. If coppicing is to continue, chestnut stools must be 'thinned' using brushwood killers, stump removal or premature cutting to prevent rapid re-growth. If the stand is to be promoted to high forest, felling and 'singling' stools can be used to create space for other species already present, or group felling and restocking practised. The overall diversity of different species groups using the canopy should increase in proportion to the greater variety of host species and the more diverse canopy structure.
Re-introduce or increase numbers of standard trees	Increases structural diversity at a stand scale, resulting in more niches for invertebrates and foraging birds, especially hole-nesters and branch feeders. Standard trees also introduce older growth elements that benefit the species requiring these stages: some retained veterans will extend the age range further (see minimum intervention below). The greater the density of the standards, the less productive the coppice will be beneath. Overdominance of the upper storey may reduce the benefits of a two-tiered structure, requiring a constant balance to be maintained in the allocation of basal area to each structural element.
Change the overall forest structure	A diversity of forest structure may be achieved by altering rotation lengths in different areas or compartments, increasing the overall number of growth stages present on site. In small forest areas this may not be feasible if the units containing each growth stage fall below a critical size, say 0.5ha. In general, greater diversity of structure will be beneficial to a wider range of species. However, where notable species are present an altered regime affecting compartment size or rotation age may be detrimental.
Operate a minimum intervention policy	Encourages dominance of chestnut in the canopy in the medium term (eg 50 years), but in the long term the stands should diversify as other trees and shrubs enter through canopy gaps. Produces abundant dead wood for fungi, saproxylic insects and lichens. Minimum intervention policies may reduce or eliminate populations of specialists requiring young growth stages and open ground.

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Appendix 1 – Summary minutes of the sweet chestnut workshop

Summary minutes of the workshop:

The honorary native: the significance of sweet chestnut in woodland conservation management

Held at: **Imperial College, Wye, Kent**
On: **Thursday 20 February 2003**

Delegates

Martin	Allison	Royal Society for the Protection of Birds (RSPB)
Jeremy	Ashworth	Esus Forestry & Woodlands Ltd
John	Badmin	Kent Field Club
Nicola	Bannister	Consultant
David	Blakesley	Horticulture Research International
Charles	Dawes	Country Landowners Association
Sally	Evans	Kent County Council
David	Gardner	Kent Butterfly Conservation
Ian	Ferguson	
Jonathan	Harding	Forestry Commission
Sue	Harris	English Heritage
Pauline	Harvey	English Nature
Ruth	Howell	Ecological Consultant
Andrew	Jones	Kent County Council
Tom	La Dell	Tom La Dell Landscape Architects
John	Leigh-Pemberton	Torry Hill
Dai	Lewis	Tillhill
Caroline	Lingham	Sevenoaks District Council
David	Maylam	English Nature
Patrick	McKernan	SE AONB
Ian	Rickards	C/o Kent Wildlife Trust
Geoffrey	Roberts	Owner
Dave	Rogers	English Nature Kent Team
Nick	Sandford	Country Landowners Association
Shirley	Thompson	Kent Bat Group
John	Tucker	Woodland Trust
Brian	Watmough	Canterbury City Council
Trevor	White	National Trust
Matthew	Woodcock	Forestry Commission

Speakers

Debbie	Bartlett	Kent County Council
Peter	Buckley	Imperial College Wye
Ralph	Harmer	Forestry Commission
Keith	Kirby	English Nature
Dave	Rossney	ESUS Forestry & Woodlands Ltd
Karen	Russell	Horticulture Research International
Michael	Walter	Royal Society for the Protection of Birds (RSPB)

Agenda

The primary purpose of this workshop is to gather views, information and opinions from participants on the conservation value of sweet chestnut woodland. We will be asking a number of questions and seeking answers from a knowledgeable audience.

Does chestnut really deserve its 'honorary' native species reputation? As a species still exploited for coppice markets, its value for butterflies, migrant birds and ephemeral ground flora is well established with conservationists. But what sort of woodland has chestnut replaced, and was that much 'better' for wildlife? If coppice markets continue to decline, is there still a case for keeping it? Would it survive non-intervention policies, or could it be successfully integrated into high forest management?

In the interests of nature conservation, should we continue to coppice uneconomic chestnut woodlands? Is it desirable or even possible to restore chestnut plantations to near-native woodland on ancient woodland sites? What practical habitat restoration solutions would we like to see, and what form should future management take?

10.00 Coffee

10.30 *Introduction to the workshop*: Peter Buckley, Imperial College

10.40 *Sweet chestnut – honorary native or horrible alien?* Keith Kirby, English Nature

11.00 *CASCADE: European research on chestnut diversity to aid conservation and improvement:* Karen Russell, Horticulture Research International

11.20 *The silviculture of chestnut coppicing*: Ralph Harmer, Forestry Commission

11.40 *Key review findings – the English chestnut habitat*: Peter Buckley, Imperial College

12.00 Group discussion: *comparative values of chestnut versus other woodland for wildlife*

13.00 Buffet lunch

13.40 *Coppicing activity and markets*: Debbie Bartlett, Kent County Council and David Rossney, ESUS

14.00 *Native woodland restoration – sweet chestnut removal in the Blean*: Michael Walter, RSPB

14.20 *A pragmatic conservationist approach to chestnut*: Keith Kirby, English Nature

14.40 Discussion in groups: *Coppicing,* restoration *or conversion - alternatives and methods*

15.20 *The future*

15.30 Tea and departure

Introduction
Peter Buckley

The number of hectares of sweet chestnut has fluctuated between 19,000 and 30,000 since 1947. The area of high forest has increased and coppice decreased. The census figures are hard to interpret because the classification of forests varied. The coppice area may be underestimated. French studies show quite a large amount of reversion. Pauline Harvey reports 12000ha in Kent from the Phase 1, field-by-field habitat survey, whilst the FC census puts the figure at 5600ha.

Sweet chestnut – honorary native or horrible alien
Keith Kirby

Good native, bad alien concept, are attitudes are changing? Our landscapes are cultural and all woods managed. Humans have used the land for 3000 years at least; woodlands are not natural. The oldest ancient forest is in Poland, a 19th century hunting forest. Therefore, does the species composition matter?

Past and future naturalness
Past – Ancient woodlands valued as past natural, for cultural heritage. Species associated with ancient woodland are limited to examples with a long continuity of woodland. Future – what would happen if woodland were allowed to develop under current processes? For example, how would the grey squirrel and the alien sycamore influence woodland? Features of the future not the past. Where does sweet chestnut fit into this?

Rackham (1980) considers sweet chestnut an honorary native (also Peterken). Evelyn thought sweet chestnut was native. Prior to the 20th century, there was uncertainty as to its status. Sweet chestnut grows from seed; is found in mixed woods; there are places named after it, eg, Chesterwood in 1272, near Colchester, Essex. Chestnut is not in the pollen record, but there is chestnut pollen in Europe (although this evidence is not infallible). There are no definite pre-Roman records in Britain. Roman/medieval evidence for chestnut takes the form of charcoal. Written chestnut records date from Henry II, eg Forest of Dean. The Tortworth chestnut is an example of a very large, ancient chestnut.

Romans introduced chestnut for food (nuts) and timber. Early records for chestnut include, north Kent, the Stour estuary, Severn estuary and the New Forest. Mostly close to the coast.

Most planted stands of chestnut, with regular arrangement, are post 1850.

Other chestnut stands are of variable size and merge into different stand types. Stands of this type are found in woods in SSSI's and NCR's. In some ways chestnut behaves as a native species, like oak or hornbeam. Sweet chestnut is an archaeophyte, an ancient introduction (spp. before 1800).

Comparing sweet chestnut with sycamore, a lot of the same arguments apply, although sycamore is more aggressive. Also, beech may not be native, what is the impact of beech?

David Maylam
There was a large planting of sweet chestnut in the Blean from 1740-1850.

CASCADE: European research on chestnut diversity to aid conservation and improvement

Karen Russell

CASCADE is an EEC funded project looking at the genetic diversity of sweet chestnut and gene flow. The aim is to aid conservation and utilisation of chestnut across Europe. Also concerned with climate change. There are 11 institutes and 6 countries involved.

The chestnut study populations are in orchards, coppice, and naturalised stands. There are 78 populations, 2-3 in one location.

The objectives are to establish/ study/produce:

- ecological optima
- use
- growth
- gene flow
- evolution and migration
- sustainable populations (genetically)
- drought tolerance
- juvenile growth (artificial conditions – Sweden)
- molecular map

Two main diseases of chestnut:

- ink disease
- chestnut blight

Uses of chestnut are country specific. The non-market benefits have been surveyed in over 800 surveys in 4 countries.

English sites
There are six English sample sites. Thermatic maps have been produced for the whole of Europe.

Ink disease in the UK is rare, but the UK chestnut population is the most susceptible to ink disease. The study found that UK chestnut populations had the highest genetic variability and it can be concluded form this that chestnut was introduced at many times. Chestnuts in Turkey show most adaptive variability.

Question
What does this mean for UK chestnuts?

Karen Russell
English sites are ecologically valuable, especially for management.

Keith Kirby
The molecular approach? If you combine different data sets you get different information.

Geoffrey Roberts
Victorian nurseries spread sweet chestnut by layering.

Karen Russell
In France chestnut was propagated by grafting.

Question
Are clonal varieties available commercially?

Karen Russell
Not from the CASCADE project. There are disease resistance recommendations. The Greek population plants don't adapt well to changing conditions – maybe others could be introduced.

The silviculture of chestnut coppicing
Ralph Harmer

There has been work on coppice in general, and hazel in particular. However, much of this knowledge and practical experience has not been written down.

FC Bulletin 64 by Rollinson deals with coppice yield.

Coppice is either simple or coppice-with-standards. The 1947 records for chestnut coverage are most reliable. 20,000ha were recorded in 1947, 1/3 simple coppice, and 2/3 coppice with standards. The problem with later records is how coppice and coppice with standards have been classified.

Market example, walking sticks exported to Germany.

Very little systematic research has taken place. Most serious research started 50 years ago. There have been 7 experiments on sweet chestnut:

- 1 chemical control
- 2 yield
- 4 silviculture.

Charles Begley carried out the most detailed research.

Nothing of value was yielded from the 4 silviculture experiments. 3 were closed and 1 damaged by squirrels.

Should the way we manage stools be changed? There isn't much information to base any changes on.

Age and size effects. These are difficult to disentangle as are related to each other. Older, larger stools respond less well than smaller, younger stools.

French on chestnut regrowth. Diameter of stump and diameter of stool.

Ralph found 4% of stools died after felling. Felling close to the ground is recommended, although not for old wood. What happens with sweet chestnut? Can we change tradition? May be able to fell throughout the year. Effects of changing when we fell? Neglected stools will re-sprout if felled within the last 50 years. Very little research on this and this is unlikely to change.

David Maylam
Axes and saw are the traditional tools for felling. There is a paper by Brian Phillips in the Quarterly Journal of Forestry comparing these tools. If sawn the stems are more vigorous and if axe cut many stems are produced, but there is very little difference long term.

Ralph Harmer
Some work by the French found the opposite.

David Rossney
Stems that are cut low down are preferred as they are the best quality. Anecdotal evidence that stools cut down lower are more stable as they produce adventitious roots.

Ralph Harmer
There is nothing in the literature. The lower down cut produces better vascularisation. There are large stools near Canterbury that have been cut low down that have not regrown. Big stools may have a problem with survival.

Dave Rossney
What is meant by low and high cutting?

Ralph Harmer
Each stool is treated as an individual, can't be prescriptive.

Karen Russell
COST programme looked at felling time and durability. There may be a durability issue with summer felling.

Key review findings – the English chestnut habitat
Peter Buckley

This has been superseded by the review.

Group discussion

Comparative values of sweet chestnut versus other woodland for wildlife

Keith Kirby
One available comparative study on sweet chestnut with other woodland is that by Ovington at Bedgebury and Abbotswood.

Peter Buckley
Ovington found the flora of sweet chestnut plots similar to that of oak plots.

David Maylam
A short rotation was traditional. The longer rotation was for hop poles. Different rotation lengths were for different uses. Sweet chestnut was coppiced on a long rotation from 1750s onwards. Species adapted themselves to the longer rotation, eg, the Blean.

Jeremy Ashworth
What is the impact of sweet chestnut nuts on wildlife versus acorns or beech mast?

Ralph Harmer
Mature sweet chestnut trees flower late compared with other tree species. Evidence from lunchtime walks suggests that chestnut fruits every year, but the nuts get maggots very quickly. Seedlings are not seen very often. Gurnell's work on small mammals and fruiting is a relevant reference.

Peter Buckley
Gurnell found sweet chestnut fruits late and that the fruit are not good for dormice.

Karen Russell
Fruit production is very sensitive to competition. If tree crowns are released fruit production is higher. Fruit production increases when chestnut is stored.

Geoffrey Roberts
Is the purpose to grow sweet chestnut for nuts or coppice? In France chestnut is only grown on soils good for nuts and at different altitudes than here in England. More than half our chestnut acreage is grown in Kent and Sussex. Should we focus on the climatic differences between SE England and other areas?

Karen Russell
East and west of England are extremely different in terms of climate, but differences in chestnut growth have not been researched.

John Tucker
In the Forest of Dean chestnut does very well, so why are the east and west populations referred to as extremes?

Karen Russell
The climatic conditions of the Forest of Dean are extremely different from the east coast, but not extreme in terms of the climatic limits of sweet chestnut. The biggest differences are between the Forest of Dean and Suffolk.

John Leigh-Pemberton
In some years there is regeneration of chestnut with substantial numbers of natural seedlings in coppice. Seen these in the past 4-5 years. Natural seedlings occur after a year of coppice growth and these can be collected and if in the right place a guard is placed around them and they are allowed to gap up. Perhaps colonises grassland much like oak, in set aside for example. Nuts are distributed by an agent.

Keith Kirby
Chestnut has a large seed, so how is it spread, what is the agent?
(Debbie Bartlett note: not natural, wild boar?).

John Leigh-Pemberton
In a good season the seeds are trodden into the ground. There are 4-5 tree seedlings per square metre in 14-15 year old coppice.

Peter Buckley
Do these seedlings survive?

John Leigh-Pemberton
No, eaten by hares or shaded out. Mostly shaded out.

Michael Walter
Chestnut does not regenerate well at the Blean, although occasionally there are spectacular seedling years, such as three years ago. Most seedlings died out.

Patrick McKernan
Pollen analysis confirms sweet chestnut is a Roman introduction. Its distribution in the Forest of Dean was so rapid, no natural species could have spread so quickly. Planted widely as does not naturalise well. SE chestnut woods are 300 yrs old. Chestnut spread as a cultural species. Cultural value of chestnut to the Romans was perhaps not so high as has been suggested; chestnuts were probably of minor importance for Romans as well. The interest in chestnut is " a new thing".

Tom La Dell
Is there any reason why plantation densities vary so much? They vary anything from 1 to 1000 stools/ha.

David Maylam
Planting densities may have varied according to the use, eg, for hooping were planted 3ft apart. A longer rotation was used for the mining industry. Chestnut supports creaked when they were likely to collapse, which made them highly prized by miners. Sweet chestnut was suitable for those supports. Planting conditions were not taken into account. Planted where for 1-2 or 3-4 year rotations they were fine, but on a 35 year rotation the poles were of little use, eg, not suitable for pales.

John Leigh-Pemberton
The cost of cutting is a significant proportion of the cost of the product. Nowadays a bi-product has no value. Pure stands of chestnut to produce straight poles are preferred now.

Keith Kirby
Maybe chestnut was tried as a gap up tree when it was considered a new 'wonder' tree.

Ralph Harmer
200-year-old literature records mixed planting to improve the crop.

David Rossney
Hornbeam woods are found pure and were maintained that way for a reason.

Nicola Bannister
The Scotney Estate, Hussey family farm, Kent. Edward Hussey the 3rd requested coppice coups were oak and hornbeam planted with sweet chestnut. Hop poles grown on long rotation. Estate records give us some clue to the nature conservation value. Diaries describe the common need for hop poles.

Patrick McKernan
Dense chestnut is bad for the heath fritillary. Areas have been opened up for this species. A dense leaf mulch is bad for violets. Constant management might be the only reason for chestnut's conservation value. Management is the key to its conservation value, not the species itself.

Keith Kirby
So species effect of chestnut is negative and the management effect positive – hence woodland structure is of key importance.

Patrick McKernan
wheat grows where chestnut woodland is being opened up.

John Leigh-Pemberton
Need to assess whether to regularly coppice a species that is bad for conservation because it is being cut, or, use a species that is good for conservation but that will not get cut. Chestnut coppice is commercial and is being cut.

John Badmin
Coppice is the backdrop in which animals etc. live. The rotation cycle and gaps make the system bio-diverse – from sampling for over 20 years. Also, opening up of rides.

David Gardner
In Thornton wood, the heath fritillary was found in newly cut areas at sites with no physical connection with sites where they were previously found. (wandering females). It may not be quite so critical to have interlinked sites.

Ian Ferguson
Selection is in favour of individuals with good dispersal.

Tom La Dell
Nightjars nest in one year old sweet chestnut coppice if large enough.

Ian Ferguson
In the west country heath fritillary feeds on heath plantain, and in Kent woods it feeds on cow wheat. There are two separate populations.

David Maylam
Check the old records to see if the woods were previously heathland.

John Badmin
Sweet chestnut is species poor, eg Southwood's paper on invertebrates. Species numbers also relate to the age of the tree in the UK.

John Leigh-Pemberton
Some recent chestnut colonisers may be at the extreme of their range.

Jay Doyle
Remember, the waved carpet moth may not have been associated with woods before it became associated with chestnut.

Keith Kirby
What was it associated with before?

Matthew Woodcock
What is the value of chestnut as a veteran?

Keith Kirby
Chestnut is considered as important at some sites, eg at Croft castle. Here species otherwise on oak can use sweet chestnut.

Dave Rogers
Confirmed Keith's statement.

Patrick McKernan
The intensity of the chestnut monoculture coppice is the problem. Scattered, large sweet chestnut veterans are probably good for conservation.

Steve Holmwood
Chris Howkins reports that chestnut nuts are the second most important in value in the world.

Delegate
Nuts also make flour.

Karen Russell
On the continent there are a high number of fungal species associated with chestnut, c. 300-350 species.

Patrick McKernan
Is there a threat of sweet chestnut fungal diseases spreading to our natives, eg oak.

Karen Russell
Chestnut blight is the only current threat.

Geoffrey Roberts
Did all US populations die?

Karen Russell
No. Chestnuts were hit by chestnut blight in America and then in Europe. It is currently 30 miles from the Normandy coast and could potentially annihilate sweet chestnut in southern England. The disease crosses from *C. dentata* to *C. sativa*.

David Rogers
How serious is this threat?

Karen Russell
Chestnut blight is considered a major threat to chestnut, particularly in the Mediterranean regions. However, the threat is not so severe now as it was. UK populations are not challenged at all. It may be possible to artificially introduce resistance to blight on oak bark before it arrives.

Ralph Harmer
C. dentata has been lost as a forest tree in the US. This may happen to chestnut if blight reaches the UK.

Coppicing activity and markets
Debbie Bartlett and David Rossney

There has been a decline in coppicing due to a decline in markets. Meanwhile there are still enquiries about the availability of palings. The market for palings is limited by supply. The growers get very little income. Cutters are available, but there is no money to invest in training and machinery; funding and health and safety are key issues. The cutters need to be legally compliant. Stakeholder workshops have been run by KCC, funded in part by the European Social Fund. These addressed safety issues and were oversubscribed.

Training topics

Bench felling is recommended as this puts less strain on the hands and wrists. Help is given on completing site risk assessment forms and training in forestry first aid.

It is not know how many cutters there are, but, there are 300 names on the database. The database can't be used directly as it is not legally available. However, if growers send in the locations where cutters are required, these can be forwarded on to cutters.

Kent Coppice Survey

Information supplied by property managers. The grid reference, area and whether sweet chestnut or mixed is recorded. So far, 3 years' of survey findings indicate the amount of sweet chestnut cut each year is pretty constant, and most coppice is mixed.

David Rossney

It will be an economic and cultural loss if cutters are lost.

Question (from Ian Rickards perhaps)

What is the position on lone working?

David Rossney
For lone working in remote places a risk assessment is required and controls need to be put in place.

John Leigh-Pemberton
Mechanisation of coppicing would be a break through and would make a difference.

David Rossney
KCC have looked at this. Bench felling is a move towards possible mechanisation.

Ralph Harmer
Felling stools next to crops, how do you get to the stools without damaging your next crop?

David Rossney
The stools grow everywhere, so easier to access (?), not like a conifer.

Debbie Bartlett
Can use workplace methods to increase achievements.

Native woodland restoration
Michael Walter

The Blean (Kent) is just over 500ha of woodland with 146ha in coppice. A study of the nightingale population has indicated there are fewer nightingales in the chestnut dominated coppice and more in the birch coppice. Of the breeding scrub birds, willow warbler, whitethroat, garden warbler and nightingale, there are fewer territories per hectare in chestnut than in birch. These species are present in the chestnut but numbers more variable.

BTO have surveyed birds in north Blean (on acid soils and *c* 1800ha) and south wood, (on basic soils, *c* 700ha) in actively managed coppice. Birch cut at 8-25 years. Different recorders recorded to different standards, so results are only a guide. They found the more alkaline soils, with dense bramble, favoured scrub bird species.

It was decided to reduce the area of chestnut on the reserve; a reduction in domination, not a removal. The objective was to convert pure monocultures into mixed species coppice, allow some reversion to high forest and provide glades etc. The methods available for removing or killing sweet chestnut are, grubbing out stumps, spraying regrowth and weed wiping. Mostly used the weed wiping approach. This was a phased approach attempting to kill off 15-20% up to a maximum of 40%. Then, in four years time go back and cut the regrowth treating a proportion of the regrowth. This phased approach is softer on the landscape, it does not create a ‘desert’ of cleared/weed killed stumps. It is also possible to watch for natural regeneration and treat stools where there will be most benefit. Areas can be reseeded with seeds collected on the reserve.

Areas where cow wheat grows under sweet chestnut or a sweet chestnut mixture are being left alone to encourage the heath fritillary.

64ha of sweet chestnut have been entered into this programme. Some areas are completed as ‘chestnut reduced zones’. There is an increase in the variety of flora in these managed areas.

Geoffrey Roberts (?)
There is a greater biomass of invertebrates in mixed coppice than single species chestnut.

Patrick McKernan
What is the dividing line between heathland and cleared woodland area?

Michael Walter
Using management to control the birch and thus end up with heath.

Matthew Woodcock
RSPB are doing this in a balanced way. The priority is to keep the habitat as woodland as the designation is ancient semi-natural woodland (ASNW). This does not mean that you can't have open spaces within the woodland.

John Leigh-Pemberton (?)
What about grazing?

Michael Walter
Yes, this is a possibility if the area is of an appropriate size.

Nicola Bannister
What about woodbank, etc. Machine operations to clear areas damage earthworks. Before using machinery, did you carry out and archaeological survey?

Michael Walter
Yes, we know where they are and so avoid damaging them.

Nicola Bannister
Grubbing out old chestnut stumps, to what extent have they damaged woodland archaeology? Sweet chestnut woods near iron ore sites, grubbing out the stumps can easily damage missed archaeology.

Michael Walter
Charcoal sites on EN area, saw pits and woodbanks located.

A pragmatic conservationist approach to chestnut
Keith Kirby

What do we do with our sweet chestnut woods now?

- Maintain species diversity and communities.
- Site specific targets for structure

Three approaches:

- Long established stands mixed with other species, maintain these
- Recent with some old chestnut areas, thin to favour other species
- Dense monocultures, treat as plantations, can we restore these to more native mixtures.

Structural targets

Aim for structural variety, eg. open space, old trees. Species of open spaces are often associated with sweet chestnut coppice. Some species have survived as a result of the coppice industry. Structural complexity to make up for the low tree species variety.

The Lowland Mixed Deciduous HAP is being drafted. The work on sweet chestnut will contribute to the drafting of the action plans.

Chestnut in the future?

Chestnut is considered non-invasive, but robust. How will this be affected by climate change? What will happen to the balance between oak, sweet chestnut and beech etc.? We need a philosophy that copes better with a cultural and dynamic landscape.

Patrick McKernan
There is a lot of chestnut outside special sites. Woods at Challock, Denge and Mereworth would be good for SSSIs.

Keith Kirby
Have we got the SSSI system right? Yes, then we need to have this in mind.

Discussion in groups:
Coppicing, restoration or conversion – alternatives and methods

Jeremy Ashworth Group

BLUE GROUP

(1) TARGETING SWEET CHESTNUT

- DAYS FOR CONSERVATION MANAGEMENT
- BUT DOESN'T PRIORITISE CONSERVATION?
- NO ECOLOGICAL JUSTIFICATION – (BAP) EXCEPT AS A MEANS TO AN END (COPPICE STRUCTURE)
- FUNGI, FOOD SOURCE FOR SMALL MAMMALS (SQUIRRELS!!!)
- MAINTAINING AN INDUSTRY
  LATER GOOD FOR SPIDERS
- POTENTIAL FOR REINVASION BY NIGHTINGALES
- VETERANS FOR RAPTORS

(2) HOW TO IMPROVE BIODIVERSITY?

- ALL MANAGEMENT COSTS MONEY
- ABANDONMENT – A PERIOD OF IMPOVERISHMENT?
- THINNING TO HIGH FOREST
  - NATURAL WINDTHROW ⟶ GAPS
    SHAKE?

DEPENDS ON SITE
SELECTION OF INDIVIDUAL STEMS

- CONCENTRATE DIFFERENT STAND TYPES
  - PURE CHESTNUT BLOCKS
  - HINTERLAND OF MIXED STANDS & STDS
- CARBON SEQUESTRATION?

Debbie Bartlett Group

(1) Where should it be encouraged - * where not

- Not much new planting
  Only done to gap up existing commercial crops

- Existing areas of SC coppice are being maintained

- Species specific – waved carpet moth

- Traditional uses – socially important

- Public interest in Traditional Woodcraft

- Recreation – People like coppice, not bothered what species

- Over mature coppice liable to windthrow – public access?

(2) Biodiversity

- Support industry

- Biodversity – edge effect of woodland rides

- Wider rides – loss of crop but important for conservation purposes.

- Question only applies in non-commercial crop

- Level of Biodiversity management is site specific

Ruth Howell Group

ENCOURAGE

WHERE	**WHERE NOT**
Near markets	Other scarce habitats
Level sites	New woodland
Workforce	Browsing pressure
Historic presence	Not traditional
Good ecology now	
Meets objectives	
Cultural History	

DIVERSITY (HOW)

Less input at restock

Natural Processes (no gapping up)

Increase open areas

Planting gaps

Long term retention
Refer to historic records

Grazing

Store

Varied rotations

Sudden change

Dave Rossney Group

Must work with the owner
Encourage chestnut

- most economic value
- best quality
- good extraction conditions
- opportunity for resurgence as fencing/timber = durable untreated sustainable
- on productive sites, suitable sites/soil
- future for high quality chestnut products (eg structural/finger jointed)

discourage chestnut

- wetter sites, uneconomic
- encourage more suitable species for sustainable products

Depends of objectives

To Consider:

- soil type
- history – woodland types

How can we diversify?

Oak standards improve diversity of insects

Chestnut $\longrightarrow$ high forest
needs <u>great</u> care in selection

- good examples A consider
- bad examples

Do not store chestnut on sandy soils

Chestnut stock – originally fruit or timber?

Start singling before 10 years creating structural diversity

Appendix 2 – Increasing capacity in the coppice sector

Debbie Bartlett, Woodlands Officer, Kent County Council

Background

The counties in the SE of England are heavily wooded with a high proportion of Ancient Woodland. Until recently most of this was managed as rotational coppice providing a source of income for landowners, who regularly sold the right to cut the standing timber, and employment for both cutters and processors of coppice product.

Since the early 1990s a marked decline has been observed in the industry with negative impacts on farmer/landowner incomes and rural employment. There is increasing concern across the South East regarding the future of wildlife associated with coppice woodlands, particularly for key BAP[5] species such as butterflies (eg heath fritillary and pearl bordered fritillary) birds (nightingale, nightjar) and mammals (eg dormice). The reasons for this disruption are complex and include:

- Competition from cheap imports of timber and wood products.
- Expectations - higher wages and better working conditions attracted many younger workers out of family businesses, exacerbated by the 1987 storm during which 'quick bucks' were offered to chain saw owners.
- Decline in summer agricultural work.
- High house prices – displacing many workers who formerly would have been in tied accommodation.
- Loss of bulk market for outgrade (the Kemsley pulp mill closed in 1991 - the nearest is now Wales).
- Overheads - compliance with Health & Safety legislation, chainsaw certification, protective equipment, insurance cover and safe lone working practices.

A complicated situation exists whereby there are multiple 'drivers' or interests keen to encourage coppice cutting from landowners suffering financially to conservation organisations concerned with encouraging biodiversity. On the other hand there are large areas of unmanaged woodland, markets that are currently under-supplied (chestnut fencing) and potential new markets – particularly for wood fuel - to be exploited. The 'pinch point' is the actual harvesting of the timber and whether this can be done efficiently.

Conservation organisations can – and will - pay contractors to coppice as 'gardening for wildlife' woodland on key sites. However for the ordinary woodland owner economics remains the key driver for woodland management and there are clear gains both in terms of rural livelihoods and delivery of public goods from halting – and in the long term reversing – the decline in the coppice industry across the south east.

[5] Biodiversity: the UK Action Plan (HMSO 1994); Kent Biodiversity Action Plan (Kent BAP Steering Group 1997); With the Grain of Nature (HMSO 2002).

To address this a series of partnership projects were based in Kent over the past five years focusing on the problems – and potential – for the traditional industry, attracting funding of £144,850, these are summarized below:

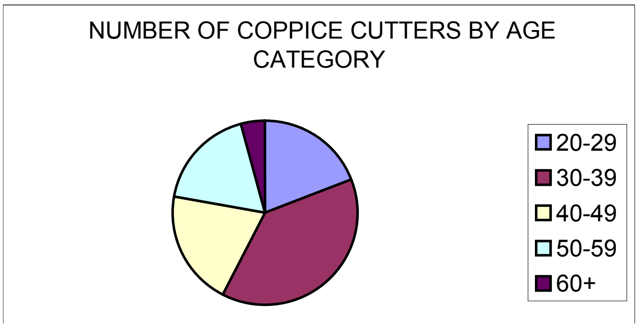
Project	Funding source
Multipurpose Woodlands in Kent/Nord Pas de Calais (1/1/99-31/12/01)	INTERREG II
Increasing Compliance with Health & Safety Legislation	European Social Fund
Increasing Compliance with Health & Safety Legislation (extension to over subscribed ESF project)	Kent Rural Revival/ SEEDA
Increasing Coppice Harvesting Efficiency	VTS (Defra)
Coppice Cutter Support Initiative	SEEDA RDA
Signs for coppice cutters	LHI

The Interreg project identified a supply problem, particularly regarding chestnut fencing products, with existing demand being greater than output. Research with the Technical Development Branch of the Forestry Commission suggested that training in harvesting techniques and work place organisation, funded by VTS (Vocational Training Scheme), could significantly increase coppice-harvesting efficiency (and consequently incomes and areas cut). However it was also established that many cutters were not compliant with the most recent Health & Safety legislation and so proficiency in use of chainsaws had to be achieved as a prerequisite.

Work on these projects has dramatically increased contact with those working in the industry – all those involved have been amazed at the number of active coppice workers that it has revealed (94 in Kent and adjacent counties) – resulting in a potentially useful contact list held in confidence. In 2003 the Coppice Cutter Support Initiative bid was developed in order to turn this into a useful database by getting permission from all involved to pass their details on to potential employers and customers. The opportunity presented by this face-to-face contact was maximised where possible by carrying out Lantra Training Needs Assessment at the same time donating to participants' coppice cutter signs[6].

The projects have shown that the majority of cutters are around the middle of their working lives, rather than as previously thought mostly in the upper age bracket and that recruitment is as likely to be in the 30s & 40s rather than solely on leaving school.

[6] 1000 of these A2 signs were produced, with funding from Nationwide, in response to requests from the cutters to "get the public off our backs" – they explain that coppice management is beneficial for wildlife to counter the negative feelings that people have about trees being cut down.



Current 'training culture' within the industry is based on paternalistic skill transfer patterns, with younger people learning directly from a small number of well-established workers who are often family members. (Out of 94, 22 (23%) were actively working with family members; an analysis of father's employment would undoubtedly emphasise this trend).

Activity is focused within small groups with little contact between them (although there is some for marketing purposes). During the TNA process (Training Needs Assessment), groups of workers sat down together and talked about the issues for the industry and their problems. It was clear that few had not given any thought to training or felt they could influence and shape their own futures. Similarly everyone who has been involved with the compliance training is encouraged to develop personal Training Action Plans.

This cohort of 94 individuals represents roughly half of the cutters currently listed on the database, held by the Forestry Commission at Bedgebury, which focuses on the former Rural Development Area in Kent, but indications are that the picture is similar across the South East. Continuing with the TNAs and delivery of the training required would contribute towards:

- Increasing wages – the cutters are on piece work, this would also make the job more attractive to new entrants.
- Increase the area of coppice cut, raising landowner incomes.
- Providing a stronger supply base and increase confidence in the processing sector, a prerequisite to product promotion and market development.
- Directly benefiting biodiversity associated with coppice management.

In conclusion, offering training is an important first step towards long-term improvements in the coppice industry. Such training would:

a. Improve the skills base in the forestry sector.
b. Improve the economic situation in farming (by increasing harvesting of farm woodlands) and forestry (employment potential of workers).
c. Improve competitiveness (of locally produced coppice products).
d. Promote production practices which maintain and enhance the landscape.
e. Protect the environment.
f. Promote forest management practices that improve the economic, ecological or social functions of forests.

The last three are of particular importance in the south-east, an area with a high proportion of ancient woodland traditionally managed as coppice and the multiple benefits of maintaining and extending this practice is identified as a priority in numerous strategic documents[7].

All training needs identified are vocational and fall within the following VTS eligible areas:

- Information and communication technology (ICT).
- Business skills.
- Marketing.
- Conservation and environmental skills.
- Diversification opportunities.
- Managing resources.
- Managing yourself and you staff.
- Technical skills (forestry).

[7] These include the SEEDA Economic strategy, the High Weald, Kent Downs and Surrey Hills AONB Management Plans, and will be a key component of the forthcoming Regional Expression of the England Forestry Strategy

Appendix 3 - List of fungi associated with sweet chestnut (SC) and pedunculate oak (PO) in the UK

(from the British Mycological Society online database records (BMSFRD) 2003)

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Abortiporus biennis (Basidiomycota: Polyporales)	SC		
Acremonium (Anamorphic fungi)		PO	
Acrospeira mirabilis (Anamorphic fungi)	SC		
Actinocladium rhodosporum (Anamorphic fungi)		PO	
Agaricus augustus (Basidiomycota: Agaricales)		PO	
Agaricus comtulus (Basidiomycota: Agaricales)		PO	
Agaricus lanipes (Basidiomycota: Agaricales)	SC		
Agaricus moelleri (Basidiomycota: Agaricales)		PO	
Agaricus placomyces (Basidiomycota: Agaricales)		PO	
Agaricus silvaticus (Basidiomycota: Agaricales)		PO	
Agaricus silvicola (Basidiomycota: Agaricales)			b
Agaricus xanthodermus (Basidiomycota: Agaricales)		PO	
Agrocybe erebia (Basidiomycota: Agaricales)		PO	
Agyriella (Anamorphic fungi)		PO	
Aleuria aurantia (Ascomycota: Pezizales)		PO	
Allophylaria basalifusca (Ascomycota: Helotiales)		PO	
Alternaria (Anamorphic fungi)	SC		
Amandinea punctata (Ascomycota: Lecanorales)		PO	
Amanita battarrae (Basidiomycota: Agaricales)		PO	
Amanita ceciliae (Basidiomycota: Agaricales)		PO	
Amanita citrina var. alba (Basidiomycota: Agaricales)			b
Amanita citrina var. citrina (Basidiomycota: Agaricales)			b
Amanita eliae (Basidiomycota: Agaricales)			b
Amanita franchetii (Basidiomycota: Agaricales)		PO	
Amanita fulva (Basidiomycota: Agaricales)			b
Amanita lividopallescens (Basidiomycota: Agaricales)		PO	
Amanita muscaria (Basidiomycota: Agaricales)			b
Amanita pantherina (Basidiomycota: Agaricales)		PO	
Amanita phalloides (Basidiomycota: Agaricales)			b
Amanita porphyria (Basidiomycota: Agaricales)		PO	
Amanita rubescens var. annulosulphurea (Basidiomycota: Agaricales)			b
Amanita rubescens var. rubescens (Basidiomycota: Agaricales)			b
Amanita spissa (Basidiomycota: Agaricales)			b
Amanita strangulata (Basidiomycota: Agaricales)		PO	
Amanita vaginata var. vaginata (Basidiomycota: Agaricales)			b
Amanita virosa (Basidiomycota: Agaricales)	SC		
Amphinema byssoides (Basidiomycota: Polyporales)	SC		
Amphiporthe leiphaemia (Ascomycota: Diaporthales)		PO	
Anavirga laxa (Anamorphic fungi)	SC		
Anguillospora crassa (Anamorphic fungi)		PO	
Anisomeridium ranunculosporum (Ascomycota: Pleosporales)		PO	
Anungitea fragilis (Anamorphic fungi)		PO	
Apiognomonia errabunda (Anamorphic fungi)		PO	
Apiognomonia errabunda (Ascomycota: Diaporthales)		PO	
Apiosporopsis carpinea (Ascomycota: Diaporthales)	SC		

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Aplanopsis terrestris (Oomycota: Saprolegniales)	SC		
Apodachlya brachynema (Oomycota: Leptomitales)		PO	
Apodachlya minima (Oomycota: Leptomitales)		PO	
Apodachlya punctata (Oomycota: Leptomitales)		PO	
Apodachlya pyrifera var. macrosporangia (Oomycota: Leptomitales)		PO	
Apodachlya seriata (Oomycota: Leptomitales)		PO	
Aposphaeria (Anamorphic fungi)		PO	
Arachnopeziza aurelia (Ascomycota: Helotiales)		PO	
Arachnopeziza eriobasis (Ascomycota: Helotiales)		PO	
Arachnoscypha aranea (Ascomycota: Helotiales)	SC		
Araiospora pulchra (Oomycota: Rhipidiales)		PO	
Arcyodes incarnata (Myxomycota: Trichiales)		PO	
Arcyria cinerea (Myxomycota: Trichiales)		PO	
Arcyria denudata (Myxomycota: Trichiales)		PO	
Arcyria incarnata (Myxomycota: Trichiales)		PO	
Arcyria nutans (Myxomycota: Trichiales)			b
Arcyria pomiformis (Myxomycota: Trichiales)		PO	
Armillaria (Basidiomycota: Agaricales)			b
Armillaria gallica (Basidiomycota: Agaricales)		PO	
Armillaria mellea (Basidiomycota: Agaricales)			b
Armillaria tabescens (Basidiomycota: Agaricales)		PO	
Arthonia radiata (Ascomycota: Arthoniales)		PO	
Arthopyrenia analepta (Ascomycota: Pleosporales)		PO	
Ascobolus lignatilis (Ascomycota: Pezizales)		PO	
Ascocoryne cylichnium (Ascomycota: Helotiales)			b
Ascocoryne sarcoides (Ascomycota: Helotiales)			b
Ascocoryne sarcoides (Ascomycota: Helotiales)		PO	
Ascodichaena rugosa (Ascomycota: Rhytismatales)		PO	
Ascomycota (Ascomycota: Incertae sedis)		PO	
Ascotremella faginea (Ascomycota: Helotiales)	SC		
Asteromella (Anamorphic fungi)	SC		
Asterophora parasitica (Basidiomycota: Agaricales)		PO	
Asterostroma laxum (Basidiomycota: Russulales)		PO	
Astraeus hygrometricus (Basidiomycota: Boletales)	SC		
Athelia (Basidiomycota: Polyporales)	SC		
Athelia decipiens (Basidiomycota: Polyporales)	SC		
Athelia epiphylla (Basidiomycota: Polyporales)	SC		
Aureobasidium pullulans (Anamorphic fungi)			b
Aureobasidium pullulans (Anamorphic fungi)		PO	
Auricularia auricula judae (Basidiomycota: Auriculariales)			b
Auricularia mesenterica (Basidiomycota: Auriculariales)		PO	
Bactrodesmium spilomeum (Anamorphic fungi)		PO	
Bactrodesmium submoniliforme (Anamorphic fungi)		PO	
Badhamia foliicola (Myxomycota: Physarales)			b
Badhamia utricularis (Myxomycota: Physarales)		PO	
Basidioradulum radula (Basidiomycota: Polyporales)		PO	
Bertia moriformis (Ascomycota: Sordariales)			b
Beverwykella pulmonaria (Anamorphic fungi)		PO	
Biscogniauxia mediterranea (Ascomycota: Xylariales)	SC		
Bispora antennata (Anamorphic fungi)		PO	
Bisporella citrina (Ascomycota: Helotiales)			b

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Bisporella fuscocincta (Ascomycota: Helotiales)		PO	
Bisporella sulfurina (Ascomycota: Helotiales)		PO	
Bjerkandera adusta (Basidiomycota: Polyporales)			b
Bjerkandera fumosa (Basidiomycota: Polyporales)			b
Bloxamia truncata (Anamorphic fungi)		PO	
Bolbitius reticulatus (Basidiomycota: Agaricales)	SC		
Boletus aereus (Basidiomycota: Boletales)		PO	
Boletus appendiculatus (Basidiomycota: Boletales)		PO	
Boletus badiorufus (Basidiomycota: Boletales)			b
Boletus badius (Basidiomycota: Boletales)			b
Boletus calopus (Basidiomycota: Boletales)		PO	
Boletus chrysenteron (Basidiomycota: Boletales)			b
Boletus citrinovirens (Basidiomycota: Boletales)		PO	
Boletus communis (Basidiomycota: Boletales)			b
Boletus edulis (Basidiomycota: Boletales)			b
Boletus erythropus (Basidiomycota: Boletales)			b
Boletus erythropus var. immutatus (Basidiomycota: Boletales)		PO	
Boletus ferrugineus (Basidiomycota: Boletales)		PO	
Boletus impolitus (Basidiomycota: Boletales)		PO	
Boletus luridiformis (Basidiomycota: Boletales)			b
Boletus luridiformis var. discolor (Basidiomycota: Boletales)			b
Boletus luridiformis var. immutatus (Basidiomycota: Boletales)		PO	
Boletus luridus (Basidiomycota: Boletales)			b
Boletus moravicus (Basidiomycota: Boletales)			b
Boletus parasiticus (Basidiomycota: Boletales)		PO	
Boletus porosporus (Basidiomycota: Boletales)			b
Boletus pruinatus (Basidiomycota: Boletales)			b
Boletus pseudoregius (Basidiomycota: Boletales)		PO	
Boletus pulverulentus (Basidiomycota: Boletales)			b
Boletus purpureus (Basidiomycota: Boletales)		PO	
Boletus queletii (Basidiomycota: Boletales)			b
Boletus radicans (Basidiomycota: Boletales)			b
Boletus reticulatus (Basidiomycota: Boletales)			b
Boletus rhodopurpureus (Basidiomycota: Boletales)		PO	
Boletus ripariellus (Basidiomycota: Boletales)		PO	
Boletus rubellus (Basidiomycota: Boletales)			b
Boletus satanas (Basidiomycota: Boletales)		PO	
Boletus satanoides (Basidiomycota: Boletales)			b
Boletus subtomentosus (Basidiomycota: Boletales)			b
Boletus xanthocyaneus (Basidiomycota: Boletales)		PO	
Botryobasidium aureum (Basidiomycota: Cantharellales)		PO	
Botryobasidium candicans (Basidiomycota: Cantharellales)	SC		
Botryobasidium danicum (Basidiomycota: Cantharellales)			b
Botryobasidium subcoronatum (Basidiomycota: Cantharellales)	SC		
Botryotinia fuckeliana (Ascomycota: Helotiales)			b
Botrytis (Anamorphic fungi)		PO	
Botrytis cinerea (Anamorphic fungi)			b
Bovista nigrescens (Basidiomycota: Agaricales)	SC		
Bovista plumbea (Basidiomycota: Agaricales)	SC		
Brachysporium bloxamii (Anamorphic fungi)			b
Brachysporium britannicum (Anamorphic fungi)			b

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Brachysporium dingleyae (Anamorphic fungi)		PO	
Brachysporium masonii (Anamorphic fungi)		PO	
Brachysporium nigrum (Anamorphic fungi)			b
Brachysporium obovatum (Anamorphic fungi)		PO	
Brevicellicium olivascens (Basidiomycota: Polyporales)	SC		
Buchwaldoboletus lignicola (Basidiomycota: Boletales)		PO	
Buellia disciformis (Ascomycota: Lecanorales)		PO	
Bulbillomyces farinosus (Basidiomycota: Polyporales)		PO	
Bulgaria inquinans (Ascomycota: Helotiales)			b
Byssocorticium atrovirens (Basidiomycota: Polyporales)	SC		
Byssocorticium efibulatum (Basidiomycota: Polyporales)		PO	
Byssomerulius corium (Basidiomycota: Polyporales)	SC		
Cacumisporium capitulatum (Anamorphic fungi)		PO	
Calcarisporium (Anamorphic fungi)		PO	
Calcarisporium arbuscula (Anamorphic fungi)		PO	
Calocera cornea (Basidiomycota: Dacrymycetales)			b
Calocera glossoides (Basidiomycota: Dacrymycetales)			b
Calocera pallidospathulata (Basidiomycota: Dacrymycetales)	SC		
Calocera viscosa (Basidiomycota: Dacrymycetales)		PO	
Calocybe carnea (Basidiomycota: Agaricales)			b
Calocybe gambosa (Basidiomycota: Agaricales)			b
Calonectria pyrochroa (Ascomycota: Hypocreales)		PO	
Calospora arausiaca (Ascomycota: Diaporthales)		PO	
Calvatia gigantea (Basidiomycota: Agaricales)		PO	
Calycella terrestris (Ascomycota: Helotiales)	SC		
Calycellina leucella (Ascomycota: Helotiales)	SC		
Calycellina punctata (Ascomycota: Helotiales)	SC		
Camarops lutea (Ascomycota: Boliniales)		PO	
Camarosporium (Anamorphic fungi)		PO	
Camarosporium oreades (Anamorphic fungi)		PO	
Camarosporium quercus (Anamorphic fungi)		PO	
Camposporium antennatum (Anamorphic fungi)		PO	
Candelabrum spinulosum (Anamorphic fungi)			b
Cantharellus cibarius var. cibarius (Basidiomycota: Cantharellales)			b
Cantharellus tubaeformis (Basidiomycota: Cantharellales)			b
Capronia pilosella (Ascomycota: Chaetothyriales)		PO	
Caudospora taleola (Ascomycota: Diaporthales)		PO	
Cephalotheca sulfurea (Ascomycota: Sordariales)		PO	
Cephalotrichum nanum (Anamorphic fungi)			b
Cephalotrichum stemonitis (Anamorphic fungi)		PO	
Ceratiomyxa fruticulosa var. fruticulosa (Myxomycota: Protosteliales)			b
Ceratocystis (Ascomycota: Microascales)		PO	
Ceratocystis paradoxa (Anamorphic fungi)		PO	
Ceratosporella stipitata (Anamorphic fungi)	SC		
Ceriporia excelsa (Basidiomycota: Polyporales)	SC		
Ceriporia reticulata (Basidiomycota: Polyporales)		PO	
Ceriporia viridans (Basidiomycota: Polyporales)			b
Ceuthospora lauri (Anamorphic fungi)	SC		
Chaetomium (Ascomycota: Sordariales)		PO	
Chaetophoma quercifolia (Anamorphic fungi)		PO	
Chaetopsis grisea (Anamorphic fungi)	SC		

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Chaetosphaerella phaeostroma (Ascomycota: Trichosphaeriales)			b
Chaetosphaeria callimorpha (Ascomycota: Sordariales)		PO	
Chaetosphaeria innumera (Ascomycota: Sordariales)		PO	
Chaetosphaeria myriocarpa (Ascomycota: Sordariales)			b
Chalara affinis (Anamorphic fungi)			b
Chalara cylindrica (Anamorphic fungi)	SC		
Chalara hughesii (Anamorphic fungi)		PO	
Chalara spiralis (Anamorphic fungi)		PO	
Chalciporus piperatus (Basidiomycota: Boletales)			b
Chlorencoelia versiformis (Ascomycota: Helotiales)		PO	
Chloridium (Anamorphic fungi)	SC		
Chloridium lignicola (Anamorphic fungi)		PO	
Chloridium virescens var. chlamydosporum (Anamorphic fungi)		PO	
Chlorociboria aeruginascens (Ascomycota: Helotiales)			b
Chlorociboria aeruginosa (Ascomycota: Helotiales)		PO	
Chlorosplenium (Ascomycota: Helotiales)		PO	
Chondrostereum purpureum (Basidiomycota: Polyporales)			b
Chromelosporium carneum (Anamorphic fungi)		PO	
Chromelosporium ochraceum (Anamorphic fungi)	SC		
Chromelosporium terrestre (Anamorphic fungi)	SC		
Chromocrea aureoviridis (Ascomycota: Hypocreales)		PO	
Ciboria (Ascomycota: Helotiales)		PO	
Ciboria americana (Ascomycota: Helotiales)	SC		
Ciboria americana (Ascomycota: Helotiales)	SC		
Ciboria batschiana (Ascomycota: Helotiales)			b
Ciborinia bresadolae (Ascomycota: Helotiales)		PO	
Ciborinia candolleana (Ascomycota: Helotiales)		PO	
Ciborinia hirtella (Ascomycota: Helotiales)			b
Cirrenalia lignicola (Anamorphic fungi)		PO	
Cistella geelmuydenii (Ascomycota: Helotiales)	SC		
Cladobotryum mycophilum (Anamorphic fungi)	SC		
Cladonia caespiticia (Ascomycota: Lecanorales)	SC		
Cladonia coniocraea (Ascomycota: Lecanorales)		PO	
Cladonia parasitica (Ascomycota: Lecanorales)		PO	
Cladosporium (Anamorphic fungi)		PO	
Cladosporium britannicum (Anamorphic fungi)		PO	
Cladosporium cladosporioides (Anamorphic fungi)	SC		
Clastoderma pachypus (Myxomycota: Echinosteliales)		PO	
Clavaria acuta (Basidiomycota: Agaricales)		PO	
Clavulina cinerea (Basidiomycota: Cantharellales)			b
Clavulina coralloides (Basidiomycota: Cantharellales)	SC		
Clavulina coralloides (Basidiomycota: Phallales)			b
Clavulina rugosa (Basidiomycota: Cantharellales)			b
Clavulinopsis corniculata (Basidiomycota: Agaricales)		PO	
Clavulinopsis helvola (Basidiomycota: Agaricales)		PO	
Clavulinopsis subtilis (Basidiomycota: Agaricales)	SC		
Cliostomum griffithii (Ascomycota: Lecanorales)		PO	
Clitocybe agrestis (Basidiomycota: Agaricales)		PO	
Clitocybe candicans (Basidiomycota: Agaricales)	SC		
Clitocybe clavipes (Basidiomycota: Agaricales)		PO	
Clitocybe costata (Basidiomycota: Agaricales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Clitocybe dealbata (Basidiomycota: Agaricales)			b
Clitocybe fragrans (Basidiomycota: Agaricales)			b
Clitocybe geotropa (Basidiomycota: Agaricales)			b
Clitocybe gibba (Basidiomycota: Agaricales)			b
Clitocybe metachroa (Basidiomycota: Agaricales)		PO	
Clitocybe nebularis (Basidiomycota: Agaricales)			b
Clitocybe odora (Basidiomycota: Agaricales)			b
Clitocybe phyllophila (Basidiomycota: Agaricales)			b
Clitocybe vibecina (Basidiomycota: Agaricales)		PO	
Clitopilus hobsonii (Basidiomycota: Agaricales)	SC		
Clitopilus prunulus (Basidiomycota: Agaricales)			b
Coccomyces dentatus (Ascomycota: Rhytismatales)			b
Coccomyces tumidus (Ascomycota: Rhytismatales)	SC		
Coemansia thaxteri (Zygomycota: Kickxellales)		PO	
Collybia acervata (Basidiomycota: Agaricales)		PO	
Collybia butyracea var. asema (Basidiomycota: Agaricales)		PO	
Collybia butyracea var. butyracea (Basidiomycota: Agaricales)	SC		
Collybia butyracea var. butyracea (Basidiomycota: Agaricales)			b
Collybia cirrhata (Basidiomycota: Agaricales)	SC		
Collybia confluens (Basidiomycota: Agaricales)			b
Collybia cookei (Basidiomycota: Agaricales)		PO	
Collybia dryophila (Basidiomycota: Agaricales)			b
Collybia erythropus (Basidiomycota: Agaricales)		PO	
Collybia erythropus (Basidiomycota: Agaricales)		PO	
Collybia fusipes (Basidiomycota: Agaricales)			b
Collybia maculata var. maculata (Basidiomycota: Agaricales)		PO	
Collybia maculata var. maculata (Basidiomycota: Agaricales)		PO	
Collybia peronata (Basidiomycota: Agaricales)			b
Collybia peronata (Basidiomycota: Agaricales)			b
Colpoma quercinum (Ascomycota: Rhytismatales)		PO	
Coltricia perennis (Basidiomycota: Hymenochaetales)	SC		
Comatricha laxa (Myxomycota: Stemonitales)		PO	
Comatricha nigra (Myxomycota: Stemonitales)			b
Comatricha tenerrima (Myxomycota: Stemonitales)		PO	
Coniella castaneicola (Anamorphic fungi)	SC		
Coniophora arida (Basidiomycota: Boletales)	SC		
Coniophora puteana (Basidiomycota: Boletales)			b
Conocybe hadrocystis (Basidiomycota: Agaricales)	SC		
Coprinus atramentarius (Basidiomycota: Agaricales)		PO	
Coprinus auricomus (Basidiomycota: Agaricales)		PO	
Coprinus comatus (Basidiomycota: Agaricales)			b
Coprinus disseminatus (Basidiomycota: Agaricales)		PO	
Coprinus domesticus (Basidiomycota: Agaricales)		PO	
Coprinus jonesii (Basidiomycota: Agaricales)	SC		
Coprinus lagopus (Basidiomycota: Agaricales)			b
Coprinus leiocephalus (Basidiomycota: Agaricales)			b
Coprinus micaceus (Basidiomycota: Agaricales)			b
Coprinus picaceus (Basidiomycota: Agaricales)		PO	
Coprinus plicatilis (Basidiomycota: Agaricales)		PO	
Coprinus xanthothrix (Basidiomycota: Agaricales)		PO	
Cordana pauciseptata (Anamorphic fungi)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Coremiella cubispora (Anamorphic fungi)		PO	
Corticium quercicola (Basidiomycota: Polyporales)		PO	
Cortinarius alboviolaceus (Basidiomycota: Agaricales)			b
Cortinarius anomalus (Basidiomycota: Agaricales)			b
Cortinarius balaustinus (Basidiomycota: Agaricales)	SC		
Cortinarius basililaceus (Basidiomycota: Agaricales)		PO	
Cortinarius cinnamomeoluteus (Basidiomycota: Agaricales)	SC		
Cortinarius claricolor (Basidiomycota: Agaricales)		PO	
Cortinarius crassus (Basidiomycota: Agaricales)		PO	
Cortinarius crocolitus (Basidiomycota: Agaricales)		PO	
Cortinarius decipiens (Basidiomycota: Agaricales)	SC		
Cortinarius decolorans (Basidiomycota: Agaricales)		PO	
Cortinarius delibutus (Basidiomycota: Agaricales)			b
Cortinarius elatior (Basidiomycota: Agaricales)		PO	
Cortinarius flexipes (Basidiomycota: Agaricales)		PO	
Cortinarius hemitrichus (Basidiomycota: Agaricales)			b
Cortinarius hinnuleus (Basidiomycota: Agaricales)		PO	
Cortinarius infractus (Basidiomycota: Agaricales)	SC		
Cortinarius melliolens (Basidiomycota: Agaricales)	SC		
Cortinarius nemorensis (Basidiomycota: Agaricales)	SC		
Cortinarius ochroleucus (Basidiomycota: Agaricales)	SC		
Cortinarius olivaceofuscus (Basidiomycota: Agaricales)	SC		
Cortinarius paleaceus (Basidiomycota: Agaricales)			b
Cortinarius pseudosalor (Basidiomycota: Agaricales)			b
Cortinarius purpurascens (Basidiomycota: Agaricales)		PO	
Cortinarius sanguineus (Basidiomycota: Agaricales)		PO	
Cortinarius semisanguineus (Basidiomycota: Agaricales)	SC		
Cortinarius torvus (Basidiomycota: Agaricales)			b
Cortinarius trivialis (Basidiomycota: Agaricales)		PO	
Cortinarius umbrinolens (Basidiomycota: Agaricales)		PO	
Cortinarius varius (Basidiomycota: Agaricales)		PO	
Corynespora biseptata (Anamorphic fungi)		PO	
Corynesporopsis quercicola (Anamorphic fungi)		PO	
Coryneum (Anamorphic fungi)		PO	
Coryneum brachyurum (Anamorphic fungi)	SC		
Coryneum japonicum (Anamorphic fungi)		PO	
Coryneum kunzei (Anamorphic fungi)			b
Craterellus cornucopioides (Basidiomycota: Cantharellales)		PO	
Craterium minutum (Myxomycota: Physarales)		PO	
Creopus gelatinosus (Ascomycota: Hypocreales)		PO	
Crepidotus cesatii var. cesatii (Basidiomycota: Agaricales)		PO	
Crepidotus epibryus (Basidiomycota: Agaricales)	SC		
Crepidotus inhonestus (Basidiomycota: Agaricales)	SC		
Crepidotus lundellii (Basidiomycota: Agaricales)	SC		
Crepidotus mollis (Basidiomycota: Agaricales)		PO	
Crepidotus variabilis (Basidiomycota: Agaricales)			b
Crocicreas coronatum (Ascomycota: Helotiales)	SC		
Crocicreas subhyalinum (Ascomycota: Helotiales)	SC		
Cryptocline cinerascens (Anamorphic fungi)		PO	
Cryptocoryneum condensatum (Anamorphic fungi)			b
Cryptodiaporthe castanea (Anamorphic fungi)	SC		

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Cryptodiaporthe castanea (Ascomycota: Diaporthales)	SC		
Cudoniella acicularis (Ascomycota: Helotiales)			b
Cudoniella clavus var. clavus (Ascomycota: Helotiales)			b
Cudoniella clavus var. grandis (Ascomycota: Helotiales)		PO	
Cyathus striatus (Basidiomycota: Agaricales)		PO	
Cylindrium (Anamorphic fungi)		PO	
Cylindrium aeruginosum (Anamorphic fungi)		PO	
Cylindrium elongatum (Anamorphic fungi)			b
Cylindrobasidium laeve (Basidiomycota: Polyporales)			b
Cystoderma (Basidiomycota: Agaricales)			b
Cystoderma amianthinum (Basidiomycota: Agaricales)			b
Cystolepiota seminuda (Basidiomycota: Agaricales)		PO	
Dacrymyces stillatus (Basidiomycota: Dacrymycetales)			b
Dactylaria obtriangularia (Anamorphic fungi)		PO	
Dactylaria purpurella (Anamorphic fungi)			b
Daedalea quercina (Basidiomycota: Polyporales)			b
Daedaleopsis confragosa (Basidiomycota: Polyporales)			b
Daedaleopsis confragosa (Basidiomycota: Polyporales)			b
Daldinia (Ascomycota: Xylariales)	SC		
Daldinia concentrica (Ascomycota: Xylariales)			b
Daldinia vernicosa (Ascomycota: Xylariales)	SC		
Datronia mollis (Basidiomycota: Polyporales)			b
Delicatula integrella (Basidiomycota: Agaricales)		PO	
Dematioscypha dematiicola (Ascomycota: Helotiales)			b
Diatrype disciformis (Ascomycota: Xylariales)			b
Diatrypella favacea (Ascomycota: Xylariales)		PO	
Diatrypella quercina (Ascomycota: Xylariales)			b
Diatrypella quercina (Ascomycota: Xylariales)		PO	
Diatrype stigma (Ascomycota: Xylariales)			b
Dictydiaethalium plumbeum (Myxomycota: Liceales)		PO	
Dictyochaeta fertilis (Anamorphic fungi)		PO	
Dictyochaeta querna (Anamorphic fungi)		PO	
Dictyochaeta simplex (Anamorphic fungi)		PO	
Diderma donkii (Myxomycota: Physarales)		PO	
Diderma effusum (Myxomycota: Physarales)		PO	
Didymium clavus (Myxomycota: Physarales)		PO	
Didymium difforme (Myxomycota: Physarales)		PO	
Didymium squamulosum (Myxomycota: Physarales)			b
Digitodesmium elegans (Anamorphic fungi)		PO	
Dimerella pineti (Ascomycota: Gyalectales)			b
Diplococcium spicatum (Anamorphic fungi)		PO	
Diplodia quercus (Anamorphic fungi)		PO	
Diplodina (Anamorphic fungi)		PO	
Diplomitoporus lindbladii (Basidiomycota: Polyporales)		PO	
Discohainesia oenotherae (Ascomycota: Helotiales)	SC		
Discosia artocreas (Anamorphic fungi)		PO	
Discosphaerina fagi (Ascomycota: Dothideales)	SC		
Ditiola peziziformis (Basidiomycota: Dacrymycetales)		PO	
Dothidotthia celtidis (Ascomycota: Pleosporales)	SC		
Dothiorella (Anamorphic fungi)		PO	
Dumontinia tuberosa (Ascomycota: Helotiales)	SC		

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Durella (Ascomycota: Helotiales)		PO	
Echinostelium brooksii (Myxomycota: Echinosteliales)		PO	
Echinostelium colliculosum (Myxomycota: Echinosteliales)		PO	
Echinostelium minutum (Myxomycota: Echinosteliales)	SC		
Eichleriella deglubens (Basidiomycota: Tremellales)		PO	
Elaphomyces granulatus (Ascomycota: Elaphomycetales)			b
Elaphomyces muricatus (Ascomycota: Elaphomycetales)	SC		
Endophragmiella biseptata (Anamorphic fungi)		PO	
Endophragmiella corticola (Anamorphic fungi)		PO	
Endophragmiella ellisii (Anamorphic fungi)	SC		
Endophragmiella fallacia (Anamorphic fungi)		PO	
Endophragmiella ovoidea (Anamorphic fungi)			b
Endophragmiella pallescens (Anamorphic fungi)		PO	
Endoxyla cirrhosa (Ascomycota: Boliniales)		PO	
Endoxyla cirrhosa (Ascomycota: Incertae sedis)		PO	
Enerthenema papillatum (Myxomycota: Stemonitales)			b
Enteridium intermedium (Myxomycota: Liceales)	SC		
Enteridium lycoperdon (Myxomycota: Liceales)			b
Enteridium olivaceum (Myxomycota: Liceales)	SC		
Enteridium splendens (Myxomycota: Liceales)		PO	
Entoloma araneosum (Basidiomycota: Agaricales)		PO	
Entoloma caccabus (Basidiomycota: Agaricales)		PO	
Entoloma cetratum (Basidiomycota: Agaricales)	SC		
Entoloma euchroum (Basidiomycota: Agaricales)	SC		
Entoloma hebes (Basidiomycota: Agaricales)		PO	
Entoloma lampropus (Basidiomycota: Agaricales)		PO	
Entoloma lividoalbum (Basidiomycota: Agaricales)		PO	
Entoloma papillatum (Basidiomycota: Agaricales)		PO	
Entoloma porphyrophaeum (Basidiomycota: Agaricales)		PO	
Entoloma rhodopolium (Basidiomycota: Agaricales)			b
Entoloma sericellum (Basidiomycota: Agaricales)			b
Epicoccum nigrum (Anamorphic fungi)		PO	
Eriopezia caesia (Ascomycota: Helotiales)		PO	
Euepixylon udum (Ascomycota: Xylariales)		PO	
Eutypella scoparia (Ascomycota: Xylariales)		PO	
Evernia prunastri (Ascomycota: Lecanorales)		PO	
Exidia glandulosa (Basidiomycota: Tremellales)			b
Exidia nucleata (Basidiomycota: Tremellales)			b
Exidia thuretiana (Basidiomycota: Tremellales)		PO	
Farlowiella carmichaeliana (Ascomycota: Hysteriales)		PO	
Fistulina hepatica (Basidiomycota: Agaricales)			b
Flammulaster granulosa (Basidiomycota: Agaricales)	SC		
Flammulina velutipes (Basidiomycota: Agaricales)			b
Flavoparmelia caperata (Ascomycota: Lecanorales)		PO	
Fomes fomentarius (Basidiomycota: Polyporales)		PO	
Fuligo candida (Myxomycota: Physarales)			b
Fuligo septica var. flava (Myxomycota: Physarales)			b
Fuligo septica var. septica (Myxomycota: Physarales)		PO	
Fusicoccum (Anamorphic fungi)		PO	
Fusicoccum castaneum (Anamorphic fungi)	SC		
Fusicoccum noxium (Anamorphic fungi)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Fusicoccum quercinum (Anamorphic fungi)		PO	
Fusidium griseum (Anamorphic fungi)			b
Galerina calyptrata (Basidiomycota: Agaricales)	SC		
Galerina marginata (Basidiomycota: Agaricales)		PO	
Ganoderma applanatum (Basidiomycota: Polyporales)			b
Ganoderma australe (Basidiomycota: Polyporales)			b
Ganoderma lucidum (Basidiomycota: Polyporales)			b
Ganoderma resinaceum (Basidiomycota: Polyporales)			b
Geastrum fimbriatum (Basidiomycota: Phallales)	SC		
Geastrum pectinatum (Basidiomycota: Phallales)		PO	
Geastrum triplex (Basidiomycota: Phallales)		PO	
Geotrichum (Anamorphic fungi)		PO	
Gibberella baccata (Anamorphic fungi)		PO	
Gibberella zeae (Anamorphic fungi)		PO	
Gloiothele lactescens (Basidiomycota: Polyporales)	SC		
Gloiothele lactescens (Basidiomycota: Russulales)	SC		
Gloniopsis praelonga (Ascomycota: Hysteriales)		PO	
Gonapodya prolifera (Chytridiomycota: Monoblepharidales)		PO	
Graddonia coracina (Ascomycota: Helotiales)		PO	
Graddonidiscus coruscatus (Ascomycota: Helotiales)			b
Graphis elegans (Ascomycota: Ostropales)		PO	
Graphis scripta (Ascomycota: Ostropales)		PO	
Graphium (Anamorphic fungi)		PO	
Graphium calicioides (Anamorphic fungi)		PO	
Grifola frondosa (Basidiomycota: Polyporales)			b
Grifola frondosa (Basidiomycota: Polyporales)			b
Guignardia aesculi (Ascomycota: Dothideales)	SC		
Guignardia punctoidea (Ascomycota: Dothideales)		PO	
Gymnopilus junonius (Basidiomycota: Agaricales)		PO	
Gymnopilus junonius (Basidiomycota: Agaricales)		PO	
Gymnopilus penetrans (Basidiomycota: Agaricales)			b
Gyroporus castaneus (Basidiomycota: Boletales)			b
Gyroporus cyanescens (Basidiomycota: Boletales)		PO	
Gyrothrix microsperma (Anamorphic fungi)	SC		
Haglundia elegantior (Ascomycota: Helotiales)	SC		
Haglundia penyardensis (Ascomycota: Helotiales)	SC		
Handkea excipuliformis (Basidiomycota: Agaricales)			b
Handkea utriformis (Basidiomycota: Agaricales)		PO	
Hapalopilus nidulans (Basidiomycota: Polyporales)			b
Haplariopsis fagicola (Anamorphic fungi)		PO	
Hebeloma (Basidiomycota: Agaricales)		PO	
Hebeloma crustuliniforme (Basidiomycota: Agaricales)			b
Hebeloma leucosarx (Basidiomycota: Agaricales)	SC		
Hebeloma longicaudum (Basidiomycota: Agaricales)		PO	
Hebeloma mesophaeum var. mesophaeum (Basidiomycota: Agaricales)			b
Hebeloma radicosum (Basidiomycota: Agaricales)		PO	
Hebeloma sacchariolens (Basidiomycota: Agaricales)			b
Hebeloma sinapizans (Basidiomycota: Agaricales)		PO	
Helicodendron paradoxum (Anamorphic fungi)		PO	
Helicogloea vestita (Basidiomycota: Incertae sedis)			b
Helicoön pluriseptatum (Anamorphic fungi)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Helicoön richonis (Anamorphic fungi)		PO	
Helicosporium (Anamorphic fungi)		PO	
Helminthosporium velutinum (Anamorphic fungi)		PO	
Helotium (Ascomycota: Helotiales)		PO	
Helvella acetabulum (Ascomycota: Pezizales)		PO	
Helvella crispa (Ascomycota: Pezizales)			b
Helvella crispa (Ascomycota: Pezizales)		PO	
Helvella elastica (Ascomycota: Pezizales)	SC		
Helvella lacunosa (Ascomycota: Pezizales)			b
Helvella macropus (Ascomycota: Pezizales)		PO	
Hemibeltrania mitrata (Anamorphic fungi)		PO	
Hemimycena candida (Basidiomycota: Agaricales)		PO	
Hemimycena cucullata (Basidiomycota: Agaricales)		PO	
Hemimycena delectabilis (Basidiomycota: Agaricales)	SC		
Hemimycena mauretanica (Basidiomycota: Agaricales)		PO	
Henicospora minor (Anamorphic fungi)		PO	
Henningsomyces candidus (Basidiomycota: Agaricales)		PO	
Hericium cirrhatum (Basidiomycota: Russulales)		PO	
Heterobasidion annosum (Basidiomycota: Russulales)			b
Hohenbuehelia reniformis (Basidiomycota: Agaricales)		PO	
Hormiactis ontariensis (Anamorphic fungi)		PO	
Hyalopeziza pygmaea (Ascomycota: Helotiales)	SC		
Hyalopeziza spinicola (Ascomycota: Helotiales)	SC		
Hyaloscypha (Ascomycota: Helotiales)		PO	
Hyaloscypha daedaleae (Ascomycota: Helotiales)	SC		
Hyaloscypha hyalina (Ascomycota: Helotiales)		PO	
Hyaloscypha zalewskii (Anamorphic fungi)			b
Hyaloscypha zalewskii (Ascomycota: Helotiales)			b
Hydnellum concrescens (Basidiomycota: Thelephorales)			b
Hydnellum scrobiculatum (Basidiomycota: Cantharellales)	SC		
Hydnellum scrobiculatum (Basidiomycota: Thelephorales)	SC		
Hydnellum spongiosipes (Basidiomycota: Thelephorales)	SC		
Hydnotrya tulasnei (Ascomycota: Pezizales)		PO	
Hydnum repandum (Basidiomycota: Cantharellales)			b
Hydnum rufescens (Basidiomycota: Cantharellales)			b
Hygrocybe colemanniana (Basidiomycota: Agaricales)	SC		
Hygrocybe psittacina var. psittacina (Basidiomycota: Agaricales)		PO	
Hygrophoropsis aurantiaca (Basidiomycota: Boletales)			b
Hygrophorus arbustivus (Basidiomycota: Agaricales)	SC		
Hygrophorus cossus (Basidiomycota: Agaricales)		PO	
Hygrophorus eburneus var. eburneus (Basidiomycota: Agaricales)		PO	
Hygrophorus nemoreus (Basidiomycota: Agaricales)		PO	
Hygrophorus persoonii (Basidiomycota: Agaricales)		PO	
Hymenochaete corrugata (Basidiomycota: Hymenochaetales)	SC		
Hymenochaete rubiginosa (Basidiomycota: Hymenochaetales)			b
Hymenochaete tabacina (Basidiomycota: Hymenochaetales)		PO	
Hymenoscyphus (Ascomycota: Helotiales)		PO	
Hymenoscyphus albidus (Ascomycota: Helotiales)		PO	
Hymenoscyphus albopunctus (Ascomycota: Helotiales)		PO	
Hymenoscyphus calyculus (Ascomycota: Helotiales)		PO	
Hymenoscyphus caudatus (Ascomycota: Helotiales)		PO	

Fungi associated	**sweet chestnut**	**pedunculate oak**	**both SC and PO**
Hymenoscyphus epiphyllus (Ascomycota: Helotiales)	SC		
Hymenoscyphus fructigenus (Ascomycota: Helotiales)			b
Hymenoscyphus humuli (Ascomycota: Helotiales)	SC		
Hymenoscyphus phyllogenus (Ascomycota: Helotiales)	SC		
Hymenoscyphus phyllophilus (Ascomycota: Helotiales)	SC		
Hymenoscyphus populneus (Ascomycota: Helotiales)	SC		
Hyphoderma cryptocallimon (Basidiomycota: Polyporales)		PO	
Hyphoderma praetermissum (Basidiomycota: Polyporales)			b
Hyphoderma puberum (Basidiomycota: Polyporales)		PO	
Hyphoderma setigerum (Basidiomycota: Polyporales)		PO	
Hyphodontia gossypina (Basidiomycota: Hymenochaetales)		PO	
Hyphodontia pallidula (Basidiomycota: Hymenochaetales)		PO	
Hyphodontia sambuci (Basidiomycota: Polyporales)		PO	
Hyphodontia subalutacea (Basidiomycota: Hymenochaetales)		PO	
Hypholoma fasciculare (Basidiomycota: Agaricales)			b
Hypholoma lateritium (Basidiomycota: Agaricales)			b
Hypholoma udum (Basidiomycota: Agaricales)	SC		
Hypochnicium punctulatum (Basidiomycota: Polyporales)			b
Hypochnicium subrigescens (Basidiomycota: Polyporales)		PO	
Hypocreaceae (Ascomycota: Hypocreales)		PO	
Hypocrea citrina (Ascomycota: Hypocreales)		PO	
Hypocrea rufa (Ascomycota: Hypocreales)		PO	
Hypoderma ilicinum (Ascomycota: Rhytismatales)	SC		
Hypogymnia physodes (Ascomycota: Lecanorales)			b
Hypospilina pustula (Ascomycota: Diaporthales)			b
Hypoxylon (Ascomycota: Xylariales)		PO	
Hypoxylon cohaerens var. microsporum (Ascomycota: Xylariales)		PO	
Hypoxylon fragiforme (Ascomycota: Xylariales)	SC		
Hypoxylon fuscum (Ascomycota: Xylariales)		PO	
Hypoxylon howeanum (Ascomycota: Xylariales)			b
Hypoxylon multiforme (Ascomycota: Xylariales)			b
Hypoxylon rubiginosum (Ascomycota: Xylariales)			b
Hypsizygus ulmarius (Basidiomycota: Agaricales)	SC		
Hysterium angustatum (Ascomycota: Hysteriales)		PO	
Hysterium pulicare (Ascomycota: Hysteriales)		PO	
Inocybe adaequata (Basidiomycota: Agaricales)			b
Inocybe assimilata (Basidiomycota: Agaricales)			b
Inocybe asterospora (Basidiomycota: Agaricales)			b
Inocybe cookei (Basidiomycota: Agaricales)		PO	
Inocybe flavella (Basidiomycota: Agaricales)	SC		
Inocybe fraudans (Basidiomycota: Agaricales)		PO	
Inocybe fuscidula var. fuscidula (Basidiomycota: Agaricales)	SC		
Inocybe geophylla var. geophylla (Basidiomycota: Agaricales)			b
Inocybe geophylla var. lilacina (Basidiomycota: Agaricales)			b
Inocybe glabripes (Basidiomycota: Agaricales)		PO	
Inocybe griseolilacina (Basidiomycota: Agaricales)		PO	
Inocybe lacera var. lacera (Basidiomycota: Agaricales)	SC		
Inocybe lanuginosa var. ovatocystis (Basidiomycota: Agaricales)	SC		
Inocybe maculata (Basidiomycota: Agaricales)			b
Inocybe napipes (Basidiomycota: Agaricales)			b
Inocybe nitidiuscula (Basidiomycota: Agaricales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Inocybe petiginosa (Basidiomycota: Agaricales)			b
Inocybe praetervisa (Basidiomycota: Agaricales)	SC		
Inocybe rimosa (Basidiomycota: Agaricales)			b
Inocybe salicis (Basidiomycota: Agaricales)	SC		
Inocybe squamata (Basidiomycota: Agaricales)			b
Inonotus dryadeus (Basidiomycota: Hymenochaetales)		PO	
Inonotus hispidus (Basidiomycota: Hymenochaetales)		PO	
Inonotus radiatus (Basidiomycota: Hymenochaetales)		PO	
Isaria umbrina (Anamorphic fungi)		PO	
Ityorhoptrum verruculosum (Anamorphic fungi)		PO	
Julella sericea (Ascomycota: Incertae sedis)		PO	
Junghuhnia nitida (Basidiomycota: Polyporales)		PO	
Kretzschmaria deusta (Ascomycota: Xylariales)			b
Kuehneromyces mutabilis (Basidiomycota: Agaricales)		PO	
Kuehneromyces mutabilis (Basidiomycota: Agaricales)		PO	
Laccaria amethystina (Basidiomycota: Agaricales)			
Laccaria laccata (Basidiomycota: Agaricales)			b
Laccaria proxima (Basidiomycota: Agaricales)		PO	
Lachnella (Basidiomycota: Agaricales)		PO	
Lachnella alboviolascens (Basidiomycota: Agaricales)		PO	
Lachnum (Ascomycota: Helotiales)	SC		
Lachnum brevipilosum (Ascomycota: Helotiales)		PO	
Lachnum capitatum (Ascomycota: Helotiales)		PO	
Lachnum castaneicola (Ascomycota: Helotiales)	SC		
Lachnum cerinum (Ascomycota: Helotiales)		PO	
Lachnum ciliare (Ascomycota: Helotiales)			b
Lachnum fuscescens var. fuscescens (Ascomycota: Helotiales)		PO	
Lachnum minutissimum (Ascomycota: Helotiales)	SC		
Lachnum niveum (Ascomycota: Helotiales)			b
Lachnum pulveraceum (Ascomycota: Helotiales)		PO	
Lachnum soppittii (Ascomycota: Helotiales)		PO	
Lachnum trapeziforme (Ascomycota: Helotiales)	SC		
Lachnum virgineum (Ascomycota: Helotiales)			b
Lacrymaria lacrymabunda (Basidiomycota: Agaricales)		PO	
Lactarius acerrimus (Basidiomycota: Russulales)		PO	
Lactarius acris (Basidiomycota: Russulales)		PO	
Lactarius aurantiacus (Basidiomycota: Russulales)		PO	
Lactarius azonites (Basidiomycota: Russulales)		PO	
Lactarius bertillonii (Basidiomycota: Russulales)		PO	
Lactarius blennius (Basidiomycota: Russulales)		PO	
Lactarius britannicus (Basidiomycota: Russulales)	SC		
Lactarius camphoratus (Basidiomycota: Russulales)			b
Lactarius chrysorrheus (Basidiomycota: Russulales)			b
Lactarius circellatus (Basidiomycota: Russulales)			b
Lactarius decipiens (Basidiomycota: Russulales)			b
Lactarius fuliginosus (Basidiomycota: Russulales)			b
Lactarius fulvissimus (Basidiomycota: Russulales)		PO	
Lactarius glyciosmus (Basidiomycota: Russulales)		PO	
Lactarius hepaticus (Basidiomycota: Russulales)	SC		
Lactarius lacunarum (Basidiomycota: Russulales)		PO	
Lactarius mairei (Basidiomycota: Russulales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Lactarius piperatus (Basidiomycota: Russulales)			b
Lactarius pterosporus (Basidiomycota: Russulales)		PO	
Lactarius quietus (Basidiomycota: Russulales)		PO	
Lactarius romagnesii (Basidiomycota: Russulales)		PO	
Lactarius rufus (Basidiomycota: Russulales)		PO	
Lactarius subdulcis (Basidiomycota: Russulales)			b
Lactarius subumbonatus (Basidiomycota: Russulales)			b
Lactarius tabidus (Basidiomycota: Russulales)			b
Lactarius torminosus (Basidiomycota: Russulales)		PO	
Lactarius turpis (Basidiomycota: Russulales)			b
Lactarius vellereus var. vellereus (Basidiomycota: Russulales)		PO	
Lactarius vietus (Basidiomycota: Russulales)		PO	
Lactarius zonarius (Basidiomycota: Russulales)		PO	
Laetiporus sulphureus (Basidiomycota: Polyporales)	SC		
Lamproderma scintillans (Myxomycota: Stemonitales)		PO	
Lanzia echinophila (Ascomycota: Helotiales)	SC		
Lanzia echinophila (Ascomycota: Helotiales)	SC		
Lanzia echinophila (Ascomycota: Incertae sedis)	SC		
Lanzia echinophila (Ascomycota: Pezizales)	SC		
Lasiosphaeria hirsuta (Ascomycota: Sordariales)		PO	
Lasiosphaeria ovina (Ascomycota: Sordariales)			b
Lasiosphaeria spermoides (Ascomycota: Sordariales)		PO	
Lecanidion atratum (Ascomycota: Patellariales)	SC		
Lecanora chlarotera (Ascomycota: Lecanorales)		PO	
Lecanora symmicta (Ascomycota: Lecanorales)		PO	
Leccinum (Basidiomycota: Boletales)		PO	
Leccinum crocipodium (Basidiomycota: Boletales)		PO	
Leccinum duriusculum (Basidiomycota: Boletales)		PO	
Leccinum populinum (Basidiomycota: Boletales)		PO	
Leccinum quercinum (Basidiomycota: Boletales)		PO	
Leccinum versipelle (Basidiomycota: Boletales)			b
Lecidella elaeochroma f. elaeochroma (Ascomycota: Lecanorales)		PO	
Lemonniera aquatica (Anamorphic fungi)		PO	
Lenzites betulinus (Basidiomycota: Polyporales)		PO	
Leocarpus fragilis (Myxomycota: Physarales)			b
Leotia lubrica (Ascomycota: Helotiales)			b
Lepiota aspera (Basidiomycota: Agaricales)			b
Lepiota castanea (Basidiomycota: Agaricales)			b
Lepiota cristata (Basidiomycota: Agaricales)			b
Lepiota echinacea (Basidiomycota: Agaricales)		PO	
Lepiota felina (Basidiomycota: Agaricales)		PO	
Lepiota helveola (Basidiomycota: Agaricales)		PO	
Lepiota ignivolvata (Basidiomycota: Agaricales)		PO	
Lepiota oreadiformis (Basidiomycota: Agaricales)		PO	
Lepiota sistrata (Basidiomycota: Agaricales)		PO	
Lepista caespitosa (Basidiomycota: Agaricales)	SC		
Lepista flaccida (Basidiomycota: Agaricales)			b
Lepista nuda (Basidiomycota: Agaricales)			b
Lepista saeva (Basidiomycota: Agaricales)		PO	
Lepraria (Anamorphic fungi)			b
Lepraria incana (Anamorphic fungi)		PO	

Fungi associated	**sweet chestnut**	**pedunculate oak**	**both SC and PO**
Leptodontidium elatius (Anamorphic fungi)		PO	
Leptosporomyces (Basidiomycota: Polyporales)	SC		
Leptothyrium castaneae (Anamorphic fungi)	SC		
Leucoagaricus badhamii (Basidiomycota: Agaricales)		PO	
Leucoagaricus georginae (Basidiomycota: Agaricales)	SC		
Leucocoprinus brebissonii (Basidiomycota: Agaricales)			b
Leucogyrophana mollusca (Basidiomycota: Boletales)	SC		
Leucogyrophana romellii (Basidiomycota: Boletales)		PO	
Lewia infectoria (Anamorphic fungi)	SC		
Libertella (Anamorphic fungi)		PO	
Licea kleistobolus (Myxomycota: Liceales)		PO	
Licea marginata (Myxomycota: Liceales)		PO	
Licea minima (Myxomycota: Liceales)	SC		
Licea operculata (Myxomycota: Liceales)	SC		
Licea parasitica (Myxomycota: Liceales)		PO	
Licea pusilla (Myxomycota: Liceales)			b
Licea pygmaea (Myxomycota: Liceales)		PO	
Limacella delicata var. vinosorubescens (Basidiomycota: Agaricales)		PO	
Lobatopedis foliicola (Anamorphic fungi)		PO	
Lophodermium petiolicola (Ascomycota: Rhytismatales)			b
Lycogala epidendrum (Myxomycota: Liceales)		PO	
Lycogala terrestre (Myxomycota: Liceales)		PO	
Lycoperdon echinatum (Basidiomycota: Agaricales)			b
Lycoperdon mammiforme (Basidiomycota: Agaricales)		PO	
Lycoperdon molle (Basidiomycota: Agaricales)		PO	
Lycoperdon nigrescens (Basidiomycota: Agaricales)			b
Lycoperdon perlatum (Basidiomycota: Agaricales)			b
Lycoperdon pyriforme (Basidiomycota: Agaricales)			b
Lyophyllum connatum (Basidiomycota: Agaricales)			b
Lyophyllum decastes (Basidiomycota: Agaricales)			b
Lyophyllum fumosum (Basidiomycota: Agaricales)	SC		
Lyophyllum infumatum (Basidiomycota: Agaricales)	SC		
Macrolepiota excoriata (Basidiomycota: Agaricales)		PO	
Macrolepiota konradii (Basidiomycota: Agaricales)		PO	
Macrolepiota mastoidea (Basidiomycota: Agaricales)		PO	
Macrolepiota procera (Basidiomycota: Agaricales)		PO	
Macrolepiota rhacodes var. rhacodes (Basidiomycota: Agaricales)		PO	
Macrotyphula fistulosa (Basidiomycota: Agaricales)		PO	
Marasmiellus candidus (Basidiomycota: Agaricales)	SC		
Marasmiellus ramealis (Basidiomycota: Agaricales)		PO	
Marasmiellus vaillantii (Basidiomycota: Agaricales)		PO	
Marasmius androsaceus (Basidiomycota: Agaricales)		PO	
Marasmius cohaerens (Basidiomycota: Agaricales)	SC		
Marasmius epiphyllus (Basidiomycota: Agaricales)			b
Marasmius oreades (Basidiomycota: Agaricales)		PO	
Marasmius quercophilus (Basidiomycota: Agaricales)		PO	
Marasmius rotula (Basidiomycota: Agaricales)		PO	
Marasmius setosus (Basidiomycota: Agaricales)		PO	
Marasmius torquescens (Basidiomycota: Agaricales)	SC		
Marasmius wynnei (Basidiomycota: Agaricales)		PO	
Megacollybia platyphylla (Basidiomycota: Agaricales)			b

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Melanelia subaurifera (Ascomycota: Lecanorales)		PO	
Melanochaeta aotearoae (Anamorphic fungi)		PO	
Melanogaster ambiguus (Basidiomycota: Boletales)			b
Melanogaster broomeianus (Basidiomycota: Boletales)		PO	
Melanoleuca exscissa var. exscissa (Basidiomycota: Agaricales)			b
Melanoleuca melaleuca var. melaleuca (Basidiomycota: Agaricales)			b
Melanoleuca polioleuca f. polioleuca (Basidiomycota: Agaricales)			b
Melanoleuca vulgaris (Basidiomycota: Agaricales)			b
Melanomma fuscidulum (Ascomycota: Pleosporales)		PO	
Melanomma pulvis pyrius (Anamorphic fungi)		PO	
Melanomma pulvis pyrius (Ascomycota: Pleosporales)		PO	
Melanopsammella preussii (Anamorphic fungi)			b
Melanopsammella vermicularioides (Anamorphic fungi)	SC		
Melanospora (Ascomycota: Hypocreales)		PO	
Melastiza scotica (Ascomycota: Pezizales)		PO	
Menispora ciliata (Anamorphic fungi)		PO	
Meripilus giganteus (Basidiomycota: Polyporales)			b
Metatrichia floriformis (Myxomycota: Trichiales)			b
Micromphale foetidum (Basidiomycota: Agaricales)		PO	
Microsphaera alphitoides (Anamorphic fungi)		PO	
Microsphaera alphitoides (Ascomycota: Erysiphales)		PO	
Microstroma album (Basidiomycota: Microstromatales)		PO	
Microthyrium ilicinum (Ascomycota: Microthyriales)		PO	
Microthyrium microscopicum (Ascomycota: Microthyriales)			b
Mirandina (Anamorphic fungi)			b
Mirandina corticola (Anamorphic fungi)		PO	
Mitrula paludosa (Ascomycota: Helotiales)		PO	
Mollisia (Ascomycota: Helotiales)	SC		
Mollisia cinerea (Ascomycota: Helotiales)			b
Mollisia cinerella (Ascomycota: Helotiales)			b
Mollisia discolor (Ascomycota: Helotiales)		PO	
Mollisia discolor var. longispora (Ascomycota: Helotiales)		PO	
Mollisia fallax (Ascomycota: Helotiales)	SC		
Mollisia heterosperma (Ascomycota: Helotiales)	SC		
Mollisia ligni (Ascomycota: Helotiales)			b
Mollisia nervicola (Ascomycota: Helotiales)	SC		
Mollisia rabenhorstii (Ascomycota: Helotiales)		PO	
Mollisia spectabilis (Ascomycota: Helotiales)		PO	
Mollisia uda (Ascomycota: Helotiales)		PO	
Mollisina acerina (Ascomycota: Helotiales)	SC		
Mollisina rubi (Ascomycota: Helotiales)		PO	
Monoblepharis (Chytridiomycota: Monoblepharidales)		PO	
Monodictys fluctuata (Anamorphic fungi)		PO	
Monodictys putredinis (Anamorphic fungi)		PO	
Morchella vulgaris (Ascomycota: Pezizales)		PO	
Mortierella verrucosa (Zygomycota: Mortierellales)		PO	
Mucilago crustacea var. crustacea (Myxomycota: Physarales)		PO	
Mucor (Zygomycota: Mucorales)		PO	
Mutinus caninus (Basidiomycota: Phallales)		PO	
Mycena acicula (Basidiomycota: Agaricales)		PO	
Mycena adonis var. adonis (Basidiomycota: Agaricales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Mycena adscendens (Basidiomycota: Agaricales)		PO	
Mycena aetites (Basidiomycota: Agaricales)		PO	
Mycena alcalina (Basidiomycota: Agaricales)		PO	
Mycena arcangeliana (Basidiomycota: Agaricales)		PO	
Mycena capillaris (Basidiomycota: Agaricales)			b
Mycena cinerella (Basidiomycota: Agaricales)		PO	
Mycena corynephora (Basidiomycota: Agaricales)		PO	
Mycena diosma (Basidiomycota: Agaricales)		PO	
Mycena epipterygia var. epipterygia (Basidiomycota: Agaricales)	SC		
Mycena erubescens (Basidiomycota: Agaricales)		PO	
Mycena filopes (Basidiomycota: Agaricales)			b
Mycena galericulata (Basidiomycota: Agaricales)			b
Mycena galopus var. candida (Basidiomycota: Agaricales)		PO	
Mycena galopus var. galopus (Basidiomycota: Agaricales)			b
Mycena galopus var. nigra (Basidiomycota: Agaricales)		PO	
Mycena haematopus (Basidiomycota: Agaricales)			b
Mycena hiemalis (Basidiomycota: Agaricales)		PO	
Mycena inclinata (Basidiomycota: Agaricales)			b
Mycena leptocephala (Basidiomycota: Agaricales)			b
Mycena longiseta (Basidiomycota: Agaricales)	SC		
Mycena maculata (Basidiomycota: Agaricales)			b
Mycena olida (Basidiomycota: Agaricales)			b
Mycena pelianthina (Basidiomycota: Agaricales)			b
Mycena polyadelpha (Basidiomycota: Agaricales)		PO	
Mycena polygramma (Basidiomycota: Agaricales)			b
Mycena pura (Basidiomycota: Agaricales)			b
Mycena rorida (Basidiomycota: Agaricales)		PO	
Mycena rosea (Basidiomycota: Agaricales)			b
Mycena sanguinolenta (Basidiomycota: Agaricales)			b
Mycena speirea (Basidiomycota: Agaricales)			b
Mycena stipata (Basidiomycota: Agaricales)	SC		
Mycena stylobates (Basidiomycota: Agaricales)			b
Mycena vitilis (Basidiomycota: Agaricales)			b
Mycoacia aurea (Basidiomycota: Polyporales)		PO	
Mycoacia fuscoatra (Basidiomycota: Polyporales)		PO	
Mycoacia uda (Basidiomycota: Polyporales)		PO	
Mycosphaerella (Ascomycota: Mycosphaerellales)			b
Mycosphaerella punctiformis (Anamorphic fungi)			b
Mycosphaerella punctiformis (Ascomycota: Mycosphaerellales)			b
Naemospora microspora (Anamorphic fungi)		PO	
Naevala perexigua (Ascomycota: Helotiales)		PO	
Naucoria bohemica (Basidiomycota: Agaricales)		PO	
Nectria (Ascomycota: Hypocreales)		PO	
Nectria cinnabarina (Anamorphic fungi)			b
Nectria cinnabarina (Ascomycota: Hypocreales)			b
Nectria coccinea (Ascomycota: Hypocreales)		PO	
Nemania bipapillata (Ascomycota: Xylariales)		PO	
Nemania confluens (Ascomycota: Xylariales)			b
Nemania serpens var. serpens (Ascomycota: Xylariales)			b
Neobulgaria pura (Ascomycota: Helotiales)		PO	
Nigrospora sphaerica (Anamorphic fungi)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Nodulisporium (Anamorphic fungi)		PO	
Oidiodendron griseum (Anamorphic fungi)	SC		
Oidiodendron tenuissimum (Anamorphic fungi)	SC		
Oliveonia pauxilla (Basidiomycota: Ceratobasidiales)	SC		
Omphalina ericetorum (Basidiomycota: Agaricales)		PO	
Opegrapha atra (Ascomycota: Arthoniales)		PO	
Opegrapha vulgata (Ascomycota: Arthoniales)		PO	
Ophiostoma piceae (Ascomycota: Microascales)	SC		
Orbilia alnea (Ascomycota: Incertae sedis)			b
Orbilia auricolor (Ascomycota: Incertae sedis)			b
Orbilia coccinella (Ascomycota: Incertae sedis)		PO	
Orbilia leucostigma (Ascomycota: Incertae sedis)		PO	
Orbilia xanthostigma (Ascomycota: Incertae sedis)		PO	
Ossicaulis lignatilis (Basidiomycota: Agaricales)	SC		
Otidea onotica (Ascomycota: Pezizales)		PO	
Oudemansiella mucida (Basidiomycota: Agaricales)		PO	
Paecilomyces (Anamorphic fungi)		PO	
Panaeolus acuminatus (Basidiomycota: Agaricales)		PO	
Panaeolus papilionaceus var. papilionaceus (Basidiomycota: Agaricales)	SC		
Panellus mitis (Basidiomycota: Agaricales)		PO	
Panellus serotinus (Basidiomycota: Agaricales)		PO	
Panellus stipticus (Basidiomycota: Agaricales)			b
Paradiacheopsis fimbriata (Myxomycota: Stemonitales)			b
Paradiacheopsis solitaria (Myxomycota: Stemonitales)			b
Parmelia saxatilis (Ascomycota: Lecanorales)		PO	
Parmelia sulcata (Ascomycota: Lecanorales)			b
Parmotrema chinense (Ascomycota: Lecanorales)			b
Paxillus involutus (Basidiomycota: Boletales)			b
Penicillium (Anamorphic fungi)		PO	
Peniophora cinerea (Basidiomycota: Russulales)		PO	
Peniophora incarnata (Basidiomycota: Russulales)			b
Peniophora lycii (Basidiomycota: Russulales)			b
Peniophora quercina (Basidiomycota: Russulales)			b
Peniophora reidii (Basidiomycota: Russulales)	SC		
Peniophora violaceolivida (Basidiomycota: Russulales)	SC		
Perenniporia fraxinea (Basidiomycota: Polyporales)		PO	
Perenniporia medulla panis (Basidiomycota: Polyporales)	SC		
Perichaena corticalis (Myxomycota: Trichiales)		PO	
Perichaena vermicularis (Myxomycota: Trichiales)	SC		
Pertusaria hymenea (Ascomycota: Pertusariales)		PO	
Pertusaria leioplaca (Ascomycota: Pertusariales)		PO	
Pestalotiopsis guepinii (Anamorphic fungi)		PO	
Peziza depressa (Ascomycota: Pezizales)		PO	
Peziza micropus (Ascomycota: Pezizales)	SC		
Peziza varia (Ascomycota: Pezizales)		PO	
Pezizella (Ascomycota: Helotiales)	SC		
Pezizella leucostigma (Ascomycota: Helotiales)	SC		
Pezizella roburnea (Ascomycota: Helotiales)			b
Pezizella vulgaris (Ascomycota: Helotiales)		PO	
Phacidiopycnis (Anamorphic fungi)		PO	
Phacidium multivalve (Anamorphic fungi)	SC		

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Phaeangella ventosa (Ascomycota: Helotiales)		PO	
Phaeohelotium umbilicatum (Ascomycota: Helotiales)	SC		
Phaeolus schweinitzii (Basidiomycota: Polyporales)			b
Phaeostalagmus cyclosporus (Anamorphic fungi)		PO	
Phaeostalagmus peregrinus (Anamorphic fungi)	SC		
Phaeostalagmus tenuissimus (Anamorphic fungi)			b
Phaeotrichosphaeria britannica (Anamorphic fungi)		PO	
Phallus impudicus (Basidiomycota: Phallales)			b
Phanerochaete laevis (Basidiomycota: Polyporales)		PO	
Phanerochaete leprosa (Basidiomycota: Russulales)	SC		
Phanerochaete velutina (Basidiomycota: Polyporales)		PO	
Phellinus ferreus (Basidiomycota: Hymenochaetales)			b
Phellinus ferruginosus (Basidiomycota: Hymenochaetales)			b
Phellinus robustus (Basidiomycota: Hymenochaetales)		PO	
Phellinus torulosus (Basidiomycota: Hymenochaetales)			b
Phellodon confluens (Basidiomycota: Thelephorales)	SC		
Phellodon melaleucus (Basidiomycota: Thelephorales)			b
Phellodon niger (Basidiomycota: Thelephorales)	SC		
Phialina lachnobrachya (Ascomycota: Helotiales)	SC		
Phialina pseudopuberula (Ascomycota: Helotiales)			b
Phialocephala fumosa (Anamorphic fungi)	SC		
Phialocephala truncata (Anamorphic fungi)		PO	
Phialophora (Anamorphic fungi)		PO	
Phialophora fastigiata (Anamorphic fungi)		PO	
Phlebia livida (Basidiomycota: Polyporales)		PO	
Phlebia radiata (Basidiomycota: Polyporales)			b
Phlebia rufa (Basidiomycota: Polyporales)		PO	
Phlebia tremellosa (Basidiomycota: Polyporales)		PO	
Phlebiella fibrillosa (Basidiomycota: Polyporales)		PO	
Phlebiella pseudotsugae (Basidiomycota: Polyporales)			b
Phlebiella sulphurea (Basidiomycota: Polyporales)			b
Phleogena faginea (Basidiomycota: Atractiellales)		PO	
Pholiota squarrosa (Basidiomycota: Agaricales)		PO	
Pholiota tuberculosa (Basidiomycota: Agaricales)		PO	
Phoma cava (Anamorphic fungi)		PO	
Phoma macrostoma (Anamorphic fungi)	SC		
Phomopsis (Anamorphic fungi)			b
Phomopsis glandicola (Anamorphic fungi)		PO	
Phomopsis quercella (Anamorphic fungi)		PO	
Phylloporus rhodoxanthus (Basidiomycota: Boletales)			b
Physarum bitectum (Myxomycota: Physarales)	SC		
Physarum cinereum (Myxomycota: Physarales)		PO	
Physarum leucophaeum (Myxomycota: Physarales)		PO	
Physarum nutans (Myxomycota: Physarales)		PO	
Physarum robustum (Myxomycota: Physarales)		PO	
Physarum scoticum (Myxomycota: Physarales)	SC		
Physcia adscendens (Ascomycota: Lecanorales)		PO	
Physcia aipolia (Ascomycota: Lecanorales)		PO	
Physisporinus vitreus (Basidiomycota: Polyporales)	SC		
Pilidium acerinum (Anamorphic fungi)			b
Piptocephalis fimbriata (Zygomycota: Zoopagales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Piptoporus quercinus (Basidiomycota: Polyporales)		PO	
Pleurophoma (Anamorphic fungi)		PO	
Pleurotheciopsis pusilla (Anamorphic fungi)	SC		
Pleurothecium recurvatum (Anamorphic fungi)		PO	
Pleurotus cornucopiae (Basidiomycota: Agaricales)		PO	
Pleurotus dryinus (Basidiomycota: Agaricales)		PO	
Pleurotus ostreatus (Basidiomycota: Agaricales)		PO	
Pluteus atromarginatus (Basidiomycota: Agaricales)		PO	
Pluteus cervinus (Basidiomycota: Agaricales)			b
Pluteus chrysophaeus (Basidiomycota: Agaricales)		PO	
Pluteus ephebeus (Basidiomycota: Agaricales)		PO	
Pluteus leoninus (Basidiomycota: Agaricales)			b
Pluteus petasatus (Basidiomycota: Agaricales)			b
Pluteus phlebophorus (Basidiomycota: Agaricales)		PO	
Pluteus romellii (Basidiomycota: Agaricales)		PO	
Pluteus salicinus (Basidiomycota: Agaricales)		PO	
Pluteus thomsonii (Basidiomycota: Agaricales)		PO	
Pluteus umbrosus (Basidiomycota: Agaricales)			b
Pocheina rosea (Acrasiomycota: Acrasiales)			b
Poculum firmum (Ascomycota: Helotiales)		PO	
Poculum firmum (Ascomycota: Incertae sedis)		PO	
Poculum petiolorum (Ascomycota: Helotiales)		PO	
Poculum sydowianum (Ascomycota: Helotiales)			b
Podoscypha multizonata (Basidiomycota: Polyporales)		PO	
Polydesmia pruinosa (Ascomycota: Helotiales)	SC		
Polyporus brumalis (Basidiomycota: Polyporales)		PO	
Polyporus durus (Basidiomycota: Polyporales)			b
Polyporus leptocephalus (Basidiomycota: Polyporales)		PO	
Polyporus squamosus (Basidiomycota: Polyporales)			b
Polyporus tuberaster (Basidiomycota: Polyporales)		PO	
Polyscytalum fecundissimum (Anamorphic fungi)			b
Porina leptalea (Ascomycota: Trichotheliales)		PO	
Postia caesia (Basidiomycota: Polyporales)			b
Postia subcaesia (Basidiomycota: Polyporales)			b
Propolis farinosa (Ascomycota: Rhytismatales)		PO	
Psathyrella artemisiae (Basidiomycota: Agaricales)	SC		
Psathyrella bipellis (Basidiomycota: Agaricales)		PO	
Psathyrella candolleana (Basidiomycota: Agaricales)			b
Psathyrella conopilus (Basidiomycota: Agaricales)		PO	
Psathyrella corrugis (Basidiomycota: Agaricales)			b
Psathyrella cotonea (Basidiomycota: Agaricales)	SC		
Psathyrella laevissima (Basidiomycota: Agaricales)			b
Psathyrella multipedata (Basidiomycota: Agaricales)		PO	
Psathyrella piluliformis (Basidiomycota: Agaricales)			b
Pseudocraterellus sinuosus (Basidiomycota: Cantharellales)			b
Pseudomicrodochium aciculare (Anamorphic fungi)	SC		
Pseudomicrodochium cylindricum (Anamorphic fungi)	SC		
Pseudospiropes obclavatus (Anamorphic fungi)		PO	
Pseudospiropes simplex (Anamorphic fungi)		PO	
Pseudotomentella mucidula (Basidiomycota: Thelephorales)	SC		
Pseudovalsa longipes (Anamorphic fungi)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Pseudovalsa longipes (Ascomycota: Diaporthales)		PO	
Pseudovalsa modonia (Anamorphic fungi)	SC		
Pseudovalsa modonia (Ascomycota: Diaporthales)	SC		
Pseudovalsaria foedans (Ascomycota: Boliniales)	SC		
Pseudovalsaria foedans (Ascomycota: Diaporthales)	SC		
Pseudovalsa umbonata (Ascomycota: Diaporthales)		PO	
Psilachnum auranticolor (Ascomycota: Helotiales)	SC		
Pulveroboletus gentilis (Basidiomycota: Boletales)		PO	
Pyricularia (Anamorphic fungi)		PO	
Pyronema omphalodes (Anamorphic fungi)		PO	
Pythium paroecandrum (Oomycota: Pythiales)	SC		
Quercus robur var. brutei, Vuilleminia comedens (Basidiomycota: Polyporales)		PO	
Radulomyces confluens (Basidiomycota: Polyporales)		PO	
Radulomyces molaris (Basidiomycota: Polyporales)		PO	
Radulomyces molaris (Basidiomycota: Polyporales)		PO	
Ramalina farinacea (Ascomycota: Lecanorales)		PO	
Ramaria stricta (Basidiomycota: Phallales)			b
Ramariopsis kunzei (Basidiomycota: Agaricales)	SC		
Resupinatus trichotis (Basidiomycota: Agaricales)		PO	
Rhabdospora acantophila (Anamorphic fungi)	SC		
Rhamphoria pyriformis (Ascomycota: Incertae sedis)		PO	
Rhinocladiella (Anamorphic fungi)		PO	
Rhipidium parthenosporum (Oomycota: Rhipidiales)		PO	
Rhizodiscina lignyota (Ascomycota: Patellariales)			b
Rhodotus palmatus (Basidiomycota: Agaricales)	SC		
Rickenella fibula (Basidiomycota: Agaricales)			b
Rigidoporus ulmarius (Basidiomycota: Polyporales)	SC		
Rosellinia aquila (Ascomycota: Xylariales)		PO	
Rozites caperatus (Basidiomycota: Agaricales)		PO	
Rubinoboletus rubinus (Basidiomycota: Boletales)			b
Russula (Basidiomycota: Russulales)	SC		
Russula aeruginea (Basidiomycota: Russulales)		PO	
Russula albonigra (Basidiomycota: Russulales)		PO	
Russula amoena (Basidiomycota: Russulales)		PO	
Russula amoenolens (Basidiomycota: Russulales)			b
Russula anthracina (Basidiomycota: Russulales)		PO	
Russula atropurpurea (Basidiomycota: Russulales)			b
Russula barlae (Basidiomycota: Russulales)	SC		
Russula betularum (Basidiomycota: Russulales)	SC		
Russula brunneoviolacea (Basidiomycota: Russulales)		PO	
Russula chloroides (Basidiomycota: Russulales)		PO	
Russula claroflava (Basidiomycota: Russulales)		PO	
Russula cyanoxantha (Basidiomycota: Russulales)			b
Russula cyanoxantha f. peltereaui (Basidiomycota: Russulales)			b
Russula delica (Basidiomycota: Russulales)			b
Russula densifolia (Basidiomycota: Russulales)			b
Russula farinipes (Basidiomycota: Russulales)		PO	
Russula fellea (Basidiomycota: Russulales)			b
Russula foetens (Basidiomycota: Russulales)			b
Russula fragilis (Basidiomycota: Russulales)			b
Russula grata (Basidiomycota: Russulales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Russula graveolens (Basidiomycota: Russulales)		PO	
Russula grisea (Basidiomycota: Russulales)		PO	
Russula heterophylla (Basidiomycota: Russulales)			b
Russula insignis (Basidiomycota: Russulales)		PO	
Russula ionochlora (Basidiomycota: Russulales)			b
Russula laeta (Basidiomycota: Russulales)		PO	
Russula lilacea (Basidiomycota: Russulales)		PO	
Russula lutea (Basidiomycota: Russulales)			b
Russula luteotacta (Basidiomycota: Russulales)		PO	
Russula melliolens (Basidiomycota: Russulales)		PO	
Russula melzeri (Basidiomycota: Russulales)	SC		
Russula nigricans (Basidiomycota: Russulales)			b
Russula nitida (Basidiomycota: Russulales)		PO	
Russula nobilis (Basidiomycota: Russulales)		PO	
Russula ochroleuca (Basidiomycota: Russulales)			b
Russula odorata (Basidiomycota: Russulales)		PO	
Russula parazurea (Basidiomycota: Russulales)			b
Russula pectinatoides (Basidiomycota: Russulales)			b
Russula pseudointegra (Basidiomycota: Russulales)			b
Russula puellaris (Basidiomycota: Russulales)		PO	
Russula queletii (Basidiomycota: Russulales)		PO	
Russula risigallina (Basidiomycota: Russulales)		PO	
Russula rosea (Basidiomycota: Russulales)		PO	
Russula sardonia (Basidiomycota: Russulales)	SC		
Russula silvestris (Basidiomycota: Russulales)	SC		
Russula silvestris (Basidiomycota: Russulales)		PO	
Russula sororia (Basidiomycota: Russulales)			b
Russula subfoetens (Basidiomycota: Russulales)		PO	
Russula velenovskyi (Basidiomycota: Russulales)		PO	
Russula vesca (Basidiomycota: Russulales)		PO	
Russula veternosa (Basidiomycota: Russulales)		PO	
Russula violeipes (Basidiomycota: Russulales)		PO	
Russula virescens (Basidiomycota: Russulales)		PO	
Russula xerampelina var. xerampelina (Basidiomycota: Russulales)		PO	
Saprolegnia hypogyna (Oomycota: Saprolegniales)		PO	
Sarcodon scabrosus (Basidiomycota: Thelephorales)			b
Schizophyllum commune (Basidiomycota: Agaricales)		PO	
Schizopora paradoxa (Basidiomycota: Hymenochaetales)			b
Schizopora paradoxa (Basidiomycota: Polyporales)	SC		
Scleroderma areolatum (Basidiomycota: Boletales)			b
Scleroderma bovista (Basidiomycota: Boletales)			b
Scleroderma cepa (Basidiomycota: Boletales)		PO	
Scleroderma citrinum (Basidiomycota: Boletales)			b
Scleroderma verrucosum (Basidiomycota: Boletales)			b
Scolecobasidium echinophilum (Anamorphic fungi)	SC		
Scoliciosporum chlorococcum (Ascomycota: Lecanorales)		PO	
Scopinella barbata (Ascomycota: Sordariales)		PO	
Scopuloides hydnoides (Basidiomycota: Russulales)			b
Scutellinia (Ascomycota: Pezizales)		PO	
Scutellinia armatospora (Ascomycota: Pezizales)		PO	
Scutellinia scutellata (Ascomycota: Pezizales)			b

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Scutellinia umbrorum (Ascomycota: Pezizales)		PO	
Scytalidium (Anamorphic fungi)		PO	
Sebacina incrustans (Basidiomycota: Tremellales)		PO	
Septoria castaneicola (Anamorphic fungi)	SC		
Septoria quercicola (Anamorphic fungi)		PO	
Septoria quercina (Anamorphic fungi)		PO	
Serpula himantioides (Basidiomycota: Boletales)			b
Simocybe centunculus var. centunculus (Basidiomycota: Agaricales)	SC		
Simocybe haustellaris (Basidiomycota: Agaricales)		PO	
Simocybe sumptuosa (Basidiomycota: Agaricales)			b
Sistotrema (Basidiomycota: Polyporales)		PO	
Sistotrema brinkmannii (Basidiomycota: Polyporales)	SC		
Skeletocutis lenis (Basidiomycota: Polyporales)	SC		
Skeletocutis nivea (Basidiomycota: Polyporales)			b
Sordaria macrospora (Ascomycota: Sordariales)		PO	
Spadicoides atra (Anamorphic fungi)		PO	
Spadicoides grovei (Anamorphic fungi)		PO	
Sparassis spathulata (Basidiomycota: Polyporales)	SC		
Sphaerobolus stellatus (Basidiomycota: Phallales)	SC		
Sphaeronaema (Anamorphic fungi)		PO	
Sphaeropsis (Anamorphic fungi)		PO	
Spirosphaera floriformis (Anamorphic fungi)		PO	
Spirosphaera minuta (Anamorphic fungi)		PO	
Sporidesmiella hyalosperma (Anamorphic fungi)		PO	
Sporidesmiella hyalosperma var. hyalosperma (Anamorphic fungi)		PO	
Sporidesmium folliculatum (Anamorphic fungi)		PO	
Sporidesmium goidanichii (Anamorphic fungi)		PO	
Sporotrichum (Anamorphic fungi)		PO	
Stachylidium (Anamorphic fungi)		PO	
Stemonitis foliicola (Myxomycota: Stemonitales)		PO	
Stemonitis fusca var. fusca (Myxomycota: Stemonitales)		PO	
Stemonitopsis amoena (Myxomycota: Stemonitales)		PO	
Stemonitopsis typhina (Myxomycota: Stemonitales)		PO	
Stereum complicatum (Basidiomycota: Russulales)		PO	
Stereum gausapatum (Basidiomycota: Russulales)			b
Stereum hirsutum (Basidiomycota: Russulales)			b
Stereum ochraceoflavum (Basidiomycota: Russulales)		PO	
Stereum rameale (Basidiomycota: Russulales)			b
Stereum rugosum (Basidiomycota: Russulales)			b
Stereum subtomentosum (Basidiomycota: Russulales)		PO	
Strobilomyces strobilaceus (Basidiomycota: Boletales)		PO	
Stropharia aeruginosa (Basidiomycota: Agaricales)		PO	
Stropharia caerulea (Basidiomycota: Agaricales)			b
Stropharia semiglobata (Basidiomycota: Agaricales)		PO	
Stropharia squamosa (Basidiomycota: Agaricales)		PO	
Stypella subhyalina (Basidiomycota: Tremellales)		PO	
Subulicystidium longisporum (Basidiomycota: Polyporales)		PO	
Suillus luteus (Basidiomycota: Boletales)		PO	
Suillus variegatus (Basidiomycota: Boletales)	SC		
Sympodiella (Anamorphic fungi)		PO	
Sympodiella foliicola (Anamorphic fungi)		PO	

Fungi associated	**sweet chestnut**	**pedunculate oak**	**both SC and PO**
Taeniolella (Anamorphic fungi)		PO	
Taeniolella stilbospora (Anamorphic fungi)		PO	
Taeniolina scripta (Anamorphic fungi)		PO	
Tapesia fusca (Ascomycota: Helotiales)		PO	
Tapesia lividofusca (Ascomycota: Helotiales)		PO	
Taphrina caerulescens (Ascomycota: Taphrinales)		PO	
Tapinella panuoides (Basidiomycota: Boletales)	SC		
Tarzetta catinus (Ascomycota: Pezizales)		PO	
Tephrocybe rancida (Basidiomycota: Agaricales)			b
Thelephora penicillata (Basidiomycota: Thelephorales)	SC		
Tomentella (Basidiomycota: Thelephorales)		PO	
Trametes gibbosa (Basidiomycota: Polyporales)			b
Trametes hirsuta (Basidiomycota: Polyporales)			b
Trametes suaveolens (Basidiomycota: Polyporales)		PO	
Trametes versicolor (Basidiomycota: Hymenochaetales)	SC		
Trametes versicolor (Basidiomycota: Polyporales)			b
Trechispora cohaerens (Basidiomycota: Polyporales)		PO	
Trechispora farinacea (Basidiomycota: Cantharellales)	SC		
Trechispora mollusca (Basidiomycota: Polyporales)	SC		
Trechispora nivea (Basidiomycota: Polyporales)		PO	
Tremella foliacea (Basidiomycota: Tremellales)		PO	
Tremella mesenterica (Basidiomycota: Tremellales)		PO	
Tremella polyporina (Basidiomycota: Tremellales)		PO	
Tremella steidleri (Basidiomycota: Tremellales)	SC		
Tricellula aquatica (Anamorphic fungi)	SC		
Tricellula ornithomorpha (Anamorphic fungi)		PO	
Trichia affinis (Myxomycota: Trichiales)		PO	
Trichia botrytis var. botrytis (Myxomycota: Trichiales)		PO	
Trichia botrytis var. cerifera (Myxomycota: Trichiales)		PO	
Trichia contorta var. contorta (Myxomycota: Trichiales)		PO	
Trichia decipiens var. decipiens (Myxomycota: Trichiales)		PO	
Trichia persimilis (Myxomycota: Trichiales)		PO	
Trichia scabra (Myxomycota: Trichiales)		PO	
Trichia varia (Myxomycota: Trichiales)		PO	
Trichoderma (Anamorphic fungi)		PO	
Tricholoma acerbum (Basidiomycota: Agaricales)			b
Tricholoma album (Basidiomycota: Agaricales)		PO	
Tricholoma argyraceum (Basidiomycota: Agaricales)		PO	
Tricholoma atrosquamosum (Basidiomycota: Agaricales)		PO	
Tricholoma basirubens (Basidiomycota: Agaricales)	SC		
Tricholoma bufonium (Basidiomycota: Agaricales)	SC		
Tricholoma cingulatum (Basidiomycota: Agaricales)	SC		
Tricholoma columbetta (Basidiomycota: Agaricales)			b
Tricholoma fulvum (Basidiomycota: Agaricales)		PO	
Tricholoma lascivum (Basidiomycota: Agaricales)		PO	
Tricholoma saponaceum var. saponaceum (Basidiomycota: Agaricales)			b
Tricholoma scalpturatum (Basidiomycota: Agaricales)		PO	
Tricholoma sejunctum (Basidiomycota: Agaricales)		PO	
Tricholoma sulphureum (Basidiomycota: Agaricales)			b
Tricholoma terreum (Basidiomycota: Agaricales)		PO	
Tricholoma ustale (Basidiomycota: Agaricales)		PO	

Fungi associated	sweet chestnut	pedunculate oak	both SC and PO
Tricholoma virgatum (Basidiomycota: Agaricales)		PO	
Tricholoma viridifucatum (Basidiomycota: Agaricales)		PO	
Tricholomopsis rutilans (Basidiomycota: Agaricales)	SC		
Tricladium castaneicola (Anamorphic fungi)	SC		
Trimmatostroma betulinum (Anamorphic fungi)		PO	
Tubaria conspersa (Basidiomycota: Agaricales)			b
Tubaria furfuracea (Basidiomycota: Agaricales)			b
Tubaria hiemalis (Basidiomycota: Agaricales)		PO	
Tubeufia (Ascomycota: Pleosporales)		PO	
Tubeufia cerea (Ascomycota: Pleosporales)			b
Tubifera ferruginosa (Myxomycota: Liceales)			b
Tulasnella thelephorea (Basidiomycota: Tulasnellales)		PO	
Tylopilus felleus (Basidiomycota: Boletales)			b
Typhula erythropus (Basidiomycota: Agaricales)		PO	
Typhula phacorrhiza (Basidiomycota: Agaricales)			b
Tyromyces (Basidiomycota: Polyporales)		PO	
Tyromyces chioneus (Basidiomycota: Polyporales)		PO	
Usnea cornuta (Ascomycota: Lecanorales)		PO	
Usnea rubicunda (Ascomycota: Lecanorales)		PO	
Valsa ambiens (Anamorphic fungi)			b
Valsa ceratosperma (Anamorphic fungi)			b
Valsa ceratosperma (Ascomycota: Diaporthales)			b
Vascellum pratense (Basidiomycota: Agaricales)		PO	
Veronaea (Anamorphic fungi)		PO	
Verticillium (Anamorphic fungi)		PO	
Virgariella ovoidea (Anamorphic fungi)		PO	
Volvariella murinella (Basidiomycota: Agaricales)	SC		
Vuilleminia comedens (Basidiomycota: Polyporales)			b
Xanthoria parietina (Ascomycota: Teloschistales)		PO	
Xanthoria polycarpa (Ascomycota: Teloschistales)		PO	
Xerula radicata (Basidiomycota: Agaricales)			b
Xylaria hypoxylon (Ascomycota: Xylariales)			b
Xylaria polymorpha (Ascomycota: Xylariales)			b
No. associated only with named species	226	672	352
Total associated	578	1024	

Glossary

allelopathic compounds substances produced by plants that limit the growth of other plants
ancient woodland a site continuously wooded for at least the past 400 years
canopy closure the growth stage of the woodland after cutting at which a continuous leaf canopy forms over the forest floor
clear felling the felling of moderate to large areas (eg >1 ha) of woodland, often involving replanting rather than natural regeneration or coppice regrowth
coppice the cutting of stems of young trees or shrubs close to the ground, causing them to resprout and to re-establish the canopy: or an area so treated
coppice with standards a system of woodland management in which timber sized trees are grown over coppice or **underwood** crop
coupe a felled or coppiced are of woodland
diversity the number and variety of plant and animal species or taxa present within a given area, dependent upon prevailing natural environmental conditions or management
early successional species a species dependent on, or favouring conditions present in the young forest growth stages following felling
gap formation
glade an open area within a woodland, usually grassy and maintained permanently by grazing
group felling the felling of small groups of trees (eg 0.1-0.5 ha in extent) at varying time intervals, creating an unevenaged forest structure
high forest a forest management system which allows the trees to grow to at least two-thirds of their full height, as opposed to earlier cutting or coppicing, producing **underwood**
late successional species a species dependent upon, or favouring areas of mature forest growth or old forest stands
mor a type of soil organic matter which remains on the surface and in which L, F and H layers can often be distinguished (L layer, the top layer of soil surface organic matter: litter more or less as it fell, F layer, the middle layer of soil surface organic matter: undergoing decomposition (fermentation) but still recognizable as leaves, twigs, etc. H layer, the bottom layer of soil surface organic matter: fully decomposed (so-called humus) and no longer traceable to the original plant parts).
moder a type of soil organic matter which is well incorporated by earthworms into the mineral soil, except for a thin layer of litter which may be present for only part of the year
neglected coppice coppice uncut for several years beyond its normal rotation age, and tending to revert to high forest (see also **stored coppice**)
normal forest a forest in which all growth stages of the trees are present, allowing an even supply of wood and timber of all sized to be produced from it each year
PAR photosynthetically active radiation, ie that part of the solar radiation spectrum used by plants for photosynthesis
regeneration the re-establishment of tree cover, either from seed shed from an adjacent canopy, or from the formation of new coppice shoots
ride a broad trackway or extraction route separating two adjacent management units of woodland
rotation the period for which trees are grown before they are cut for produce
shake a defect in freshly felled timber resulting from the splitting of wood along an annual ring (ring shake) or radiating from the pith (star shake)
snag dead standing trees, stumps or large attached branches
soil-water deficit a soil moisture deficit caused by the removal of water through surface evaporation or transpiration, measured in millimetres from field capacity
stand an (often uniform) are or tract of woodland, or other type of vegetation

standard large trees, generally intended for construction timber, overstanding coppice
underwood (see coppice with standards)
stocking the density per unit are of trees used or selected for planting, natural regeneration of thinning operations
stored coppice a stand of coppice origin which has been allowed to grow on beyond its normal rotation age
thinning removal of plants, trees etc to improve the growth of those remaining
underwood general name for a wood consisting of coppice shoots, root suckers and pollard poles, grown for wood rather than timber

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Top left: Using a home-made moth trap.
Peter Wakely/English Nature 17,396
Middle left: Co_2 experiment at Roudsea Wood and Mosses NNR, Lancashire.
Peter Wakely/English Nature 21,792
Bottom left: Radio tracking a hare on Pawlett Hams, Somerset.
Paul Glendell/English Nature 23,020
Main: Identifying moths caught in a moth trap at Ham Wall NNR, Somerset.
Paul Glendell/English Nature 24,888

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