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BIODEGRADABLE POLYMERS

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1. INTRODUCTION

Biodegradable polymers are a newly emerging field. A vast number of biodegradable polymers have been synthesized recently and some microorganisms and enzymes capable of degrading them have been identified. In developing countries, environmental pollution by synthetic polymers has assumed dangerous proportions. As a result, attempts have been made to solve these problems be including biodegradability into polymers in everyday use through slight modifications of their structures.

Biodegradation is a natural process by which organic chemicals in the environment are converted to simpler compounds, mineralized and redistributed through elemental cycles such as the carbon, nitrogen and sulphur cycles. Biodegradation can only occur within the biosphere as microorganisms play a central role in the biodegradation process.

A number of standards authorities have sought to produce definitions for biodegradable plastics and some of these are provided below:

> ISO 472: 1988—A plastic designed to undergo a significant change in its chemical structure under specific environmental conditions resulting in a loss of some properties that may vary as measured by standard test methods appropriate to the plastics and application in a period of time that determines its classification. The change in chemical structure results from the action of naturally occurring microorganisms.
> ASTM sub-committee D20.96 proposal—Degradable plastics are plastic materials that undergo bond scission in the backbone of a polymer through chemical, biological and/or

physical forces in the environment at a rate which leads to fragmentation or disintegration of the plastics.

Japanese Biodegradable Plastic Society[1] draft proposal—Biodegradable plastics are polymeric materials which are changed into lower molecular weight compounds where at least one step in the degradation process is through metabolism in the presence of naturally occurring organisms.

DIN 103.2 working group on biodegradable polymers—Biodegradation of a plastic material is a process leading to naturally occurring metabolic end products.

General definition of biodegradation—It is a process whereby bacteria, fungi, yeasts and their enzymes consume a substance as a food source so that its original form disappears. Under appropriate conditions of moisture, temperature and oxygen availability, biodegradation is a relatively rapid process. Biodegradation for limited periods is a reasonable target for the complete assimilation and disappearance of an article leaving no toxic or environmentally harmful residue.

Biodegradable polymers are useful for various applications in medical, agriculture, drug release and packaging fields.

2. NATURAL BIODEGRADABLE POLYMERS

Biopolymers are polymers formed in nature during the growth cycles of all organisms; hence, they are also referred to as natural polymers. Their synthesis generally involves enzyme-catalyzed, chain growth polymerization reactions of activated monomers, which are typically formed within cells by complex metabolic processes.

2.1. *Polysaccharides*

For materials applications, the principal polysaccharides of interest are cellulose and starch, but increasing attention is being given to the more complex carbohydrate polymers produced by bacteria and fungi, especially to polysaccharides such as xanthan, curdlan, pullulan and hyaluronic acid. These latter polymers generally contain more than one type of carbohydrate unit, and in many cases these polymers have regularly arranged branched structures. Starch, for example, is a physical combination of branched and linear polymers (amylopectin and amylose, respectively), but it contains only a single type of carbohydrate, glucose.

Both cellulose and starch are composed of hundreds or thousands of D-glucopyranoside repeating units. These units are linked together by acetal bonds formed between the hemiacetal carbon atom, C_1, of the cyclic glucose structure in one unit and a hydroxyl group at either the C_3 (for cellulose and amylose) or the C_6 (for the branch units in amylopectin) atoms in the adjacent unit. This type of structure occurs because in aqueous solution, glucose can exist in either the acyclic aldehyde or cyclic hemiacetal form, and the latter form is the structure that become incorporated into the polysaccharide. Also, the cyclic form can exist as one of two isomers, the α-isomer with an axial OH group on the ring or the β-isomer with an equatorial OH group. In starch the glucopyranoside ring is present in the α-form while in cellulose the repeating units exist in the β-form. Because of this difference, enzymes that catalyze acetal hydrolysis reactions during the biodegradation of each of these two

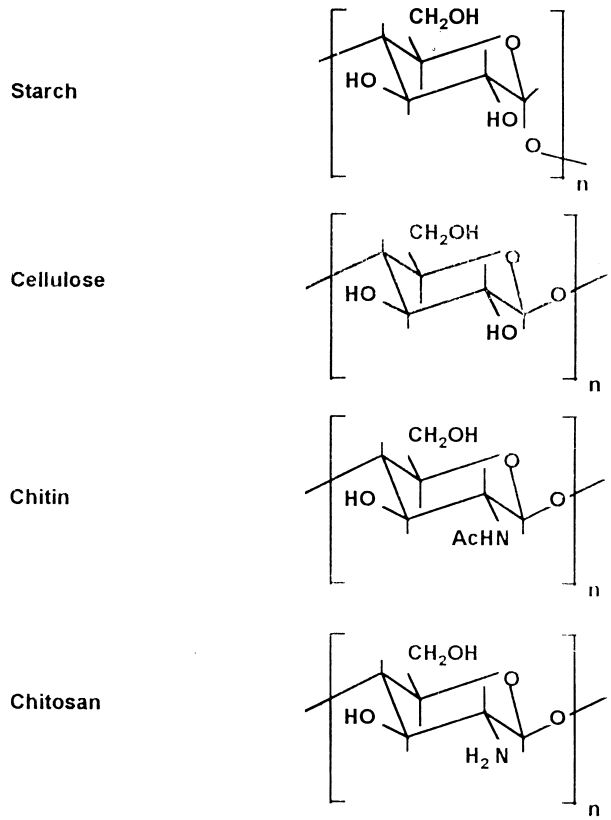


Fig. 1. Structures of polysaccharides.

polysaccharides are different and are not interchangeable. Fig. 1 shows the structures of some polysaccharides.

2.1.1. *Starch*

Starch is a polymer which occurs widely in plants. The principal crops used for its production include potatoes, corn and rice. In all of these plants, starch is produced in the form of granules, which vary in size and somewhat in composition from plant to plant. In general, the linear polymer, amylose, makes up about 20 wt% of the granule, and the branched polymer, amylopectin, the remainder. Amylose is crystalline and can have a number average molecular weight as high as 500 000, but it is soluble in boiling water. Amylopectin is insoluble in boiling water, but in their use in foods, both fractions are readily hydrolyzed at the acetal link by enzymes. The α-1,4-link in both components of starch is attacked by amylases (Fig. 2a) and the α-1,6-link in amylopectin is attacked by glucosidases.

Starch has been widely used as a raw material in film production because of increasing prices and decreasing availability of conventional film-forming resins.[2] Starch films possess low permeability and are thus attractive materials for food packaging. Starch is also useful for

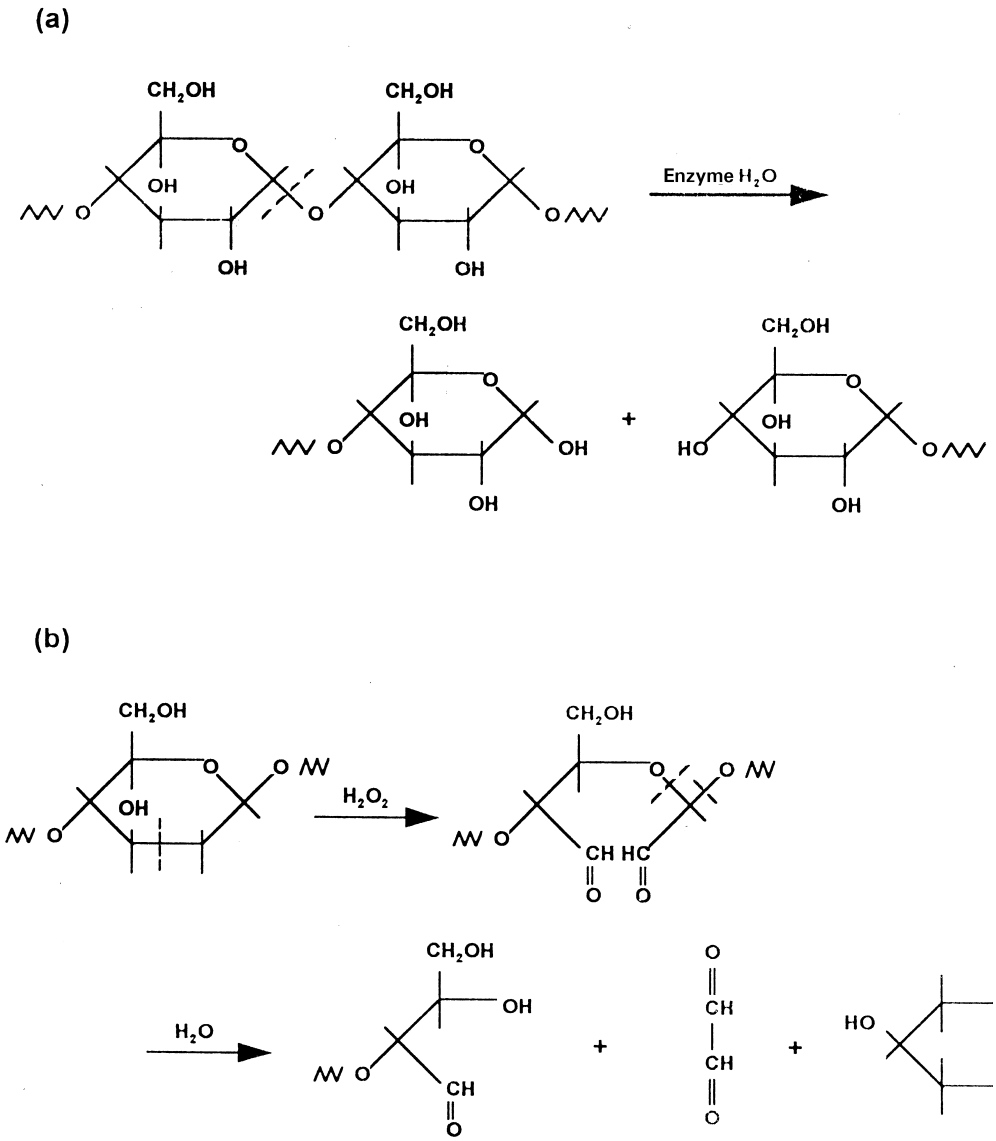


Fig. 2. Enzymatic hydrolysis of (a) starch and (b) cellulose.

making agricultural mulch films because it degrades into harmless products when placed in contact with soil microorganisms.

Research on starch includes investigation of its water adsorptive capacity, the chemical modification of the molecule, its behaviour under agitation and high temperature, and its resistance to thermomechanical shear. Although starch is a polymer, its stability under stress is not high. At temperatures higher than 150°C, the glucoside links start to break, and above 250°C the starch grain endothermally collapses. At low temperatures, a phenomenon known as retrogradation is observed. This is a reorganization of the hydrogen bonds and an aligning

of the molecular chains during cooling. In extreme cases under 10°C, precipitation is observed. Thus, though starch can be dispersed into hot water and cast as films, the above phenomenon causes brittleness in the film.

In its application in biodegradable plastics, starch is either physically mixed in with its native granules, kept intact, or melted and blended on a molecular level with the appropriate polymer. In either form, the fraction of starch in the mixture which is accessible to enzymes can be degraded by either, or both, amylases and glucosidases. The starch molecule has two important functional groups, the –OH group that is susceptible to substitution reactions and the C–O–C bond that is susceptible to chain breakage. The hydroxyl group of glucose has a nucleophilic character. By reaction of its –OH group, modification of various properties can be obtained. One example is the reaction with silane to improve its dispersion in polyethylene.[3] Crosslinking or bridging of the –OH groups changes the structure into a network while increasing the viscosity, reducing water retention and increasing its resistance to thermomechanical shear.

Acetylated starch does have several advantages as a structural fibre or film-forming polymer as compared to native starch. The acetylation of starch is a well-known reaction and is a relatively easy derivative to synthesize.[4] Starch acetate is considerably more hydrophobic than is starch and has been shown to have better retention of tensile properties in aqueous environments. Another advantage is that starch acetate has an improved solubility compared to starch and is easily cast into films from simple solvents. The degree of acetylation is easily controlled by transesterification, allowing polymers to be produced with a range of hydrophobicities. Starch has been acetylated[5] [with a high content (70%) of linear amylose] and its enzymatic degradation studied. Starch acetate was prepared by acetylation of starch with a pyridine/acetic anhydride mixture and cast into films from solutions of 90% formic acid. A series of films with a range of acetyl content were then exposed to buffered amylase solutions. It was found that with a sufficient acetyl content, the wet strength of the films was maintained in the aqueous solutions, but that the acetyl content was sufficiently low to permit degradation by a mixture of alpha and beta amylases within a period of 1 h. These films might be useful as membranes in bioreactors which could then be degraded by the addition of enzymes to the system.

Starch has been used for many years as an additive to plastic for various purposes. Starch was added as a filler[6] to various resin systems to make films that were impermeable to water but permeable to water vapour. Starch as a biodegradable filler in LDPE was reported.[7,8] A starch-filled polyethylene film was prepared[9] which becomes porous after the extraction of the starch. This porous film can be readily invaded by microorganisms and rapidly saturated with oxygen, thereby increasing polymer degradation by biological and oxidative pathways. Otey *et al.*[10] in a study on starch-based films, found that a starch–polyvinyl alcohol film could be coated with a thin layer of water-resistant polymer to give a degradable agricultural mulching film. Starch-based polyethylene films were formulated[11,12] and consisted of up to 40% starch, urea, ammonia and various portions of low-density polyethylene (LDPE) and poly(ethylene-*co*-acrylic acid) (EAA). The EAA acted as a compatibilizer, forming a complex between the starch and the PE in the presence of ammonia. The resulting blend could be cast or blown into films, and had physical properties approaching those of LDPE.

Three techniques were used to incorporate large amounts of starch as a filler into disposable polyvinyl chloride (PVC) plastics.[13] In the first technique, a starch xanthate solution

was prepared by mixing starch with aqueous NaOH and then adding a small amount of carbon disulphide (usually 0.1 mol CS_2 per mol starch). To this starch–xanthate solution, a PVC latex was added. The starch–xanthate and PVC resins were then coprecipitated by adding $NaNO_2$ and alum. The fine powder obtained from this was blended with dioctyl phthalate (DOP). In the second technique (a concentration method), whole starch was gelatinized by heating in water before mixing into the PVC latex. After removing the water, dry product was mixed with DOP. In the third method, starch was dry-blended with PVC and DOP. These films appear to be useful for a variety of agricultural applications.[14]

The possibility of chemically combining starch or starch-derived products with commercial resins in such a manner that the starch would serve as both a filler and a crosslinking agent may provide a feasible approach for incorporating starch into plastics.

Since isocyanates are highly reactive with hydroxyl groups, they can be used to prepare a number of reactive resins that crosslink with starch. The addition of starch to isocyanate resins considerably reduced costs and improved solvent resistance and strength properties.[15] Starch can be modified with nonpolar groups, such as fatty esters, before the isocyanate reaction to improve the degree of reactivity.[16] A method was developed[17] to incorporate starch as a filler and crosslinking agent in diisocyanate-modified polyesters to yield elastomers. Dosmann and Steel[18] added starch to urethane systems to yield shock-absorbing foams. Bennett *et al.*[19] reported that 10–40% of a rigid urethane foam formulation can be starch. These studies demonstrated that starch products cause foams to be more flame resistant and more readily attacked by soil microorganisms.

2.1.2. *Cellulose*

Many polymer researchers are of the opinion that polymer chemistry had its origins with the characterization of cellulose. Cellulose was isolated for the first time some 150 years ago. Cellulose differs in some respects from other polysaccharides produced by plants, the molecular chain being very long and consisting of one repeating unit (cellobiose). Naturally, it occurs in a crystalline state. From the cell walls, cellulose is isolated in microfibrils by chemical extraction. In all forms, cellulose is a very highly crystalline, high molecular weight polymer, which is infusible and insoluble in all but the most aggressive, hydrogen bond-breaking solvents such as *N*-methylmorpholine-*N*-oxide. Because of its infusibility and insolubility, cellulose is usually converted into derivatives to make it more processable.

Some fungi can secrete enzymes that catalyze oxidation reactions of either cellulose itself or the lower molecular weight oligomers produced from the enzymatic hydrolysis of cellulose. Of these, the peroxidases can provide hydrogen peroxide for free radical attack on the C_2–C_3 positions of cellulose to form ‘aldehyde’ cellulose, which is very reactive and can hydrolyze to form lower molecular weight fragments (Fig. 2b) while other oxidative enzymes can oxidize glucose and related oligomers to glucuronic acids.

Bacteria also secrete both endo- and exoenzymes, some of which form complexes that act jointly in degrading cellulose to form carbohydrate nutrients which the microorganisms utilize for survival.[20,21]

Aerobic soil environments generally contain a consortia of several different type of degrading bacteria and fungi which operate cooperatively. Primary microorganisms degrade cellulose to glucose and cellodextrins, a portion of which they utilize, and secondary

microorganisms, which provide enzymes that degrade the cellodextrins to glucose, which they consume. By consuming glucose the latter assist in the growth of the primary microorganism because they prevent the build-up of the cellodextrins, which can inhibit glucanases if they are present in the environment at high concentrations. The final products from aerobic biodegradation are ultimately CO_2 and water.

In anaerobic environments, a variety of final products are formed, including CO_2 hydrogen, methane, hydrogen sulphide and ammonia. CO_2 can be formed by oxidative reactions which utilize inorganic compounds, such as sulphate and nitrate ions, in the environment as oxidizing agents. Hydrogen produced by some anaerobic bacteria can be utilized by autotrophic bacteria to reduce oxidized compounds and CO_2 to form either acetic acid or methane.

Cellulose has received more attention than any other polymer since it is attacked by a wide variety of microorganisms, and since it is often used in textiles without additives to complicate the interpretation of results. Cellulose represents an appreciable fraction of the waste products that make up sewage and refuse. It is fortunate that it does decompose readily. Fermentation of cellulose has been suggested as a source of chemicals such as ethanol and acetic acid, but this has not achieved any commercial importance to date.

All of the important derivatives of cellulose are reaction products of one or more of the three hydroxyl groups, which are present in each glucopyranoside repeating unit, including: (1) ethers, e.g. methyl cellulose and hydroxyl-ethyl cellulose; (2) esters, e.g. cellulose acetate and cellulose xanthate, which is used as a soluble intermediate for processing cellulose into either fibre or film forms, during which the cellulose is regenerated by controlled hydrolysis; and (3) acetals, especially the cyclic acetal formed between the C_2 and C_3 hydroxyl groups and butyraldehyde.

The biodegradation of cellulose is complicated, because cellulose exists together with lignin, for example, in wood cell walls. White-rot fungi secrete exocellular peroxidases to degrade lignin preferentially and, to a lesser extent, cellulases to degrade the polysaccharides in order to produce simple sugars which serve as nutrients for these microorganisms. Brown-rot fungi secrete enzymes for the degradation of cellulose and the hemicelluloses. Soft-rot fungi, also degrade principally these two types of polysaccharides.

Cellulose esters represent a class of polymers that have the potential to participate in the carbon cycle via microbiologically catalyzed de-esterification and decomposition of the resulting cellulose and organic acids. Cellulose acetate is currently used in high volume applications ranging from fibres, to films, to injection moulding thermoplastics. It has the physical properties and relatively low material costs that have tended to exclude other biodegradable polymers from being widely accepted in the marketplace.

Gardener *et al.*[22] have developed a series of cellulose acetate films, differing in their degree of substitution, that were evaluated in this bench-scale system. In addition, commercially available biodegradable polymers such as poly(hydroxybutyrate-*co*-valerate) (PHBV) and polycaprolactone (PCL) were included as points of reference. Based on film disintegration and film weight loss, cellulose acetates, having degrees of substitution less than approximately 2.20, compost at rates comparable to that of PHBV. NMR and GPC analyses of composted films indicate that low molecular weight fractions are removed preferentially from the more highly substituted and slower degrading cellulose acetates.

Reese[23] presented evidence of esterase activity on soluble cellulose acetates with a low

degree of substitution (DS, 0.76 sites esterified per anhydroglucose monomer). A pure culture of Pestalotiopsis Westerdijkii Quarter Master (QM) 381 was reported to completely utilize this low DS cellulose ester. However, Reese did not find any evidence that the fully substituted cellulose triacetate could be biodegraded. Cantor and Mechalas[24] found evidence of esterase activity on reverse osmosis membranes composed of cellulose acetate (DS 2.5). Using infrared analysis, up to 50% deacylation was detected on the desalinating surface. No reduction in acylation was detected with cellulose triacetate. Dong Gue *et al.*[25] recently presented evidence of anaerobic biodegradation of cellulose acetate (DS 1.7) with about 9% weight loss over a 60-day period. Recently, Buchanan *et al.*[26,27] presented evidence supporting the inherent biodegradability of cellulose acetate with naturally occurring microorganisms in activated sludge and in aerobic microbial cultures.

Komarck *et al.*[28] studied biodegradation of radiolabelled cellulose acetate and cellulose propionate with a naturally derived mixed microbial culture derived from activated sludge. Radiolabelled cellulose esters were synthesized with either [1-^{14}C]-acetate or [1-^{14}C]-propionate and back hydrolyzed to the desired degree of substitution (DS) ranging from 1.77 to 2.64. Biodegradation was measured in an *in vitro* aerobic culture system that was designed to capture $^{14}CO_2$ produced by the aerobic microbial metabolism of the cellulose esters. Microorganisms were able to extensively degrade cellulose [1-^{14}C]-acetate (CA) with DS values ranging from 1.85 to 2.57 over periods of 14–31 days. More than 80% of the original ^{14}C-polymeric carbon was biodegraded to $^{14}CO_2$ for CA substrates with a DS of 1.85. CA polymers with a DS of 2.07 and 2.57 yielded over 60% conversion to $^{14}CO_2$. The amount of biodegradation that was observed for cellulose [1-^{14}C]-propionate with DS values of 2.11, 2.44 and 2.64 were lower than the corresponding acetyl ester and ranged from 0.09 to 1.1%. However, cellulose [1-^{14}C]-propionate with a DS of 1.77 and 1.84 underwent very rapid degradation in the mixed culture system, with 70–80% conversion of the labelled polymeric carbon metabolized to $^{14}CO_2$ in 29 days. The high level of microbial utilization of carbon from both cellulose esters and its conversion to CO_2 confirms the biodegradability of these polymers and the potential they have for total mineralization in natural, microbiologically active environments.

The biodegradation of cellulose ethers has been studied extensively and it is known that cellulose ethers with a DS of less than 1 will degrade due to attack of microorganisms at the unsubstituted residues of the polymers. The ether linkages on the cellulose backbone are considered resistant to microbial attack. By contrast, there have been conflicting reports concerning the biodegradation potential of cellulose esters. Stutzenberger and Kahler[29] have reported that cellulose acetate (CA) is a poor substrate, because of its extreme resistance to microbial attack. However, Reese[23] has isolated cellulolytic filtrates, which deacetylated soluble CA (DS = 0.76) and insoluble cellobiose octaacetate. Furthermore, Cantor and Mechalas[24] have demonstrated that CA reverse-osmosis membranes with a DS of 2.5 suffer losses in semipermeability due to microbial attack. These reports suggest that the synergistic action of esterase and cellulase-producing microorganisms act in concert to degrade CA. One possible mechanistic pathway would involve attack by cellulase enzymes on the unsubstituted residues in the polymer backbone. Enzymatic cleavage of the acetyls by esterase (or simple chemical hydrolysis) would then serve to expose additional unsubstituted residues, which could also be digested by the action of cellulase enzymes which further would serve eventually to degrade CA completely in the environment.

2.1.3. *Chitin and chitosan*

Chitin is a macromolecule found in the shells of crabs, lobsters, shrimps and insects. It consists of 2-acetamide-2-deoxy-β-D-glucose through the β-(1-4)-glycoside linkage. Chitin can be degraded by chitinase. Chitin fibres have been utilized for making artificial skin and absorbable sutures.[30] Chitin is insoluble in its native form but chitosan, the partly deacetylated form, is water soluble. The materials are biocompatible and have antimicrobial activities as well as the ability to absorb heavy metal ions. They also find applications in the cosmetic industry because of their water-retaining and moisturizing properties. Using chitin and chitosan as carriers, a water-soluble prodrug has been synthesized.[31]

Modified chitosans have been prepared with various chemical and biological properties.[32] *N*-Carboxymethylchitosan and *N*-carboxybutylchitosan have been prepared for use in cosmetics and in wound treatment.[33]

Chitin derivatives can also be used as drug carriers,[34] and a report of the use of chitin in absorbable sutures shows that chitins have the lowest elongation among suture materials consisting of chitin, poly(glycolic acid) (PGA), plain catgut and chromic catgut.[35] The tissue reaction of chitin is similar to that of PGA.

2.1.4. *Alginic acid*

Many polysaccharides in solution form gels upon the introduction of counterions. The degree of cross-linking is dependent on various factors such as pH, type of counterion, and the functional charge density of these polymers. Alginates have been studied extensively for their ability to form gels in the presence of divalent cations.[36–41]

Alginate is a binary linear heteropolymer containing 1,4-linked α-L-guluronic acid and β-D-mannuronic acid. Alginic acid forms water-soluble salts with monovalent cations, low molecular weight amines, and quaternary ammonium compounds. It becomes water-insoluble in the presence of polyvalent cations such as Ca^{2+}, Be^{2+}, Cu^{2+}, Al^{3+} and Fe^{3+}. Alginate gels have been used widely in controlled release drug delivery systems. Alginates have been used to encapsulate various herbicides, microorganisms and cells.

2.2. *Polypeptides of natural origin*

The proteins that have found applications as materials are, for the most part, neither soluble nor fusible without degradation, so they are used in the form in which they are found in nature. This description is especially true for the fibrous proteins wool, silk and collagen. All proteins are specific copolymers with regular arrangements of different types of α-amino acids, so the biosynthesis of proteins is an extremely complex process involving many different types of enzymes. In contrast, the enzymatic degradation of proteins, with general purpose proteases, is a relatively straightforward, amide hydrolysis reaction.

2.2.1. *Gelatin*

Gelatin, an animal protein, consists of 19 amino acids joined by peptide linkages and can be hydrolyzed by a variety of the proteolytic enzymes to yield its constituent amino acids or peptide components.[42] This nonspecificity is a desirable factor in intentional biodegradation. Gelatin is a water-soluble, biodegradable polymer with extensive industrial, pharmaceutical,

and biomedical uses, has been employed for coatings and microencapsulating various drugs,[43–49] and for preparing biodegradable hydrogels.[50–53]

A method was developed to prepare a simple, flexible gelatin film-based artificial skin that could adhere to an open wound and protect it against fluid loss and infection. The approach used was to mix polyglycerols, either as it is or after epoxizing them with epichlorohydrin, with commercially available gelatin and to cast films on teflon-covered trays.[52] The films were tough and adhered to open wounds spontaneously. They could be loaded with bioactive molecules, such as growth factors and antibiotics that would be released over several days. The films could be sterilized with γ-rays or prepared under sterile conditions.

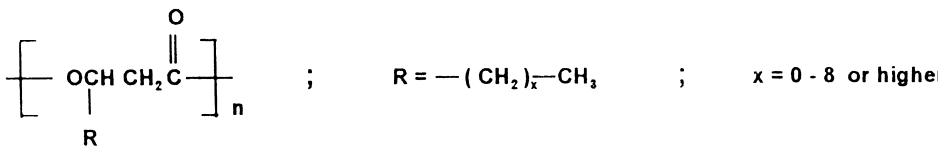
Chemical modification of natural polymers by grafting serves the twofold purpose of utilizing renewable, naturally derived products such as proteins, as replacements for petroleum-based polymers and as biodegradable compositions which can be tailored for the slower or faster rates of degradation.

In order to extend the application of grafting for the modification of natural polymers, T. Kuwajima *et al.*[54] grafted methyl methacrylate onto gelatins by radical initiators and studied these in aqueous solution at temperatures between 60 and 80°C. Among the initiators used (peroxysulphates, α,α'-azobisisobutylonitrile, and benzoyl peroxide), potassium peroxysulphate was found to be the most efficient in this particular graft polymerization. From kinetic data with this initiator, it was shown that: (1) the efficiency of grafting is higher at lower temperatures; (2) a sharp increase in the efficiency of grafting occurs at the later period of the polymerization at high temperature, which is attributable to the combination of the homopolymer and the backbone gelatin; and (3) generally, the number of branches was small and the molecular weight of the branched polymer was high in this polymerization.

Kumar *et al.*[55] prepared gelatin-g-poly(ethyl acrylate) in an aqueous medium, using $K_2S_2O_8$ as an initiator. The composition of the graft copolymer was dependent upon the temperature and duration of the reaction. The number of grafting sites was small and the molecular weight of the grafted poly(ethylacrylate) branches was high. Three copolymer samples with grafting efficiencies of 33.3, 61.0 and 84.0% were tested for their microbial susceptibility in a synthetic medium employing a mixed inoculum of *Bacillus subtilis*, *Pseudomonas aeruginosa*, and *Serratia marcescens*. The weight losses were found to be 12, 10.1 and 6.0%, respectively, after six weeks of incubation. The extent of degradation seems to decrease with increasing grafting efficiency. There was initial rapid weight loss accompanied by an exponential increase in the bacterial population and pH of the culture medium during the first week. Nitrogen analysis also showed the polymer utilization. A parallel set of experiments, carried out by employing the samples as the only source of both carbon and nitrogen, showed a marginal but definite increase in the utilization of the polymer.

2.3. *Bacterial polyesters*

The natural polyesters, which are produced by a wide variety of bacteria as intracellular reserve materials, are receiving increased attention for possible applications as biodegradable, melt processable polymers which can be produced from renewable resources. The members of this family of thermoplastic biopolymers, which have the general structure given below, can show variation in their material properties from rigid brittle plastics, to flexible plastics with good impact properties to strong tough elastomers, depending on the size of the pendant alkyl group, R, and the composition of the polymer.[56–58]



All of these polyesters contain units which are 100% optically pure at the β-position, so all are 100% isotactic. The polymer with $R = CH_3$, poly-β-hydroxybutyrate (PHB), is highly crystalline with a melting temperature of 180°C and a glass transition temperature, T_g, of approximately 5°C.[59] This combination of very high crystallinity and relatively high T_g makes the films and plastics of PHB very brittle, so copolymers with units containing other alkyl groups, especially $R = C_2H_5$, are preferred. All of these materials are inherently biodegradable. Polyesters with longer alkyl substituents, with $x = 3–6$ or so, are also produced by a variety of bacteria, generally in the form of copolymers which have lower degrees of crystallinity and lower T_m and T_g values. As a result, these longer alkyl chain polyesters are useful as thermoplastic elastomers, which can have excellent strength and toughness, and yet are also inherently biodegradable.

Considerable interest arose recently when a large-scale, controlled fermentation process was developed[60] for the production of copolymers of PHB. Feeding the bacteria with a variety of carbon sources led to the production of different copolymers and a material was obtained with better mechanical properties than PHB.[61–70]

The biodegradation of PHB and its copolymers has been studied in environments such as soil, activated sludge and sea water.[65] Films (0.07 mm thick) of PHB (homopolymer), a copolymer of 91% 3HB and 9% 4HB and a copolymer of 50% 3HB and 50% 3HV were subjected to biodegradation in soil. The fastest biodegradation rate was obtained for P(3HB-*co*-9% 4HB). In activated sludge the P(3HB-*co*-9% 4HB) was completely decomposed after two weeks.[65]

The native polyesters are also hydrolyzed in water at a very slow rate. *In vivo* this is the main degradation mechanism, involving chain scission of the polymer. The hydrolytic degradation of hydroxybutyrate–hydroxyvalerate copolymers *in vitro* begins with a surface modification, accompanied by water diffusion into the matrix.[71] A progressive increase in porosity facilitates the diffusion by removal of degradation products. Doi *et al.*[72] report that the hydrolytic degradation of microbial polyesters occurs by homogeneous erosion over two stages: random hydrolytic chain scission of the ester group leading to a decrease in molecular weight, followed by a second step ($M_n \sim 13\,000$) in which more weight loss occurs.

Bacterial polyesters have also been blended with PE and PS. The goal was to expand their physical properties while retaining biodegradability.[73,74] The biodegradation of PHB and copolymers of PHB with γ-hydroxy valerate (PHV) was monitored using an accelerated test based on a chemostate-like technique.[75] Latex films of the polyesters were compared with paper coated on the side with latex and the materials were immersed in a broth containing microorganisms isolated from activated sludge. The latex films were readily degraded and the coated papers lost about 60% of their initial weight after a week of degradation.[75]

3. POLYMERS WITH HYDROLYZABLE BACKBONES

Polymers with hydrolyzable backbones have been found to be susceptible to biodegradation. Fig. 3 shows the structures for some polymers with hydrolyzable backbones.

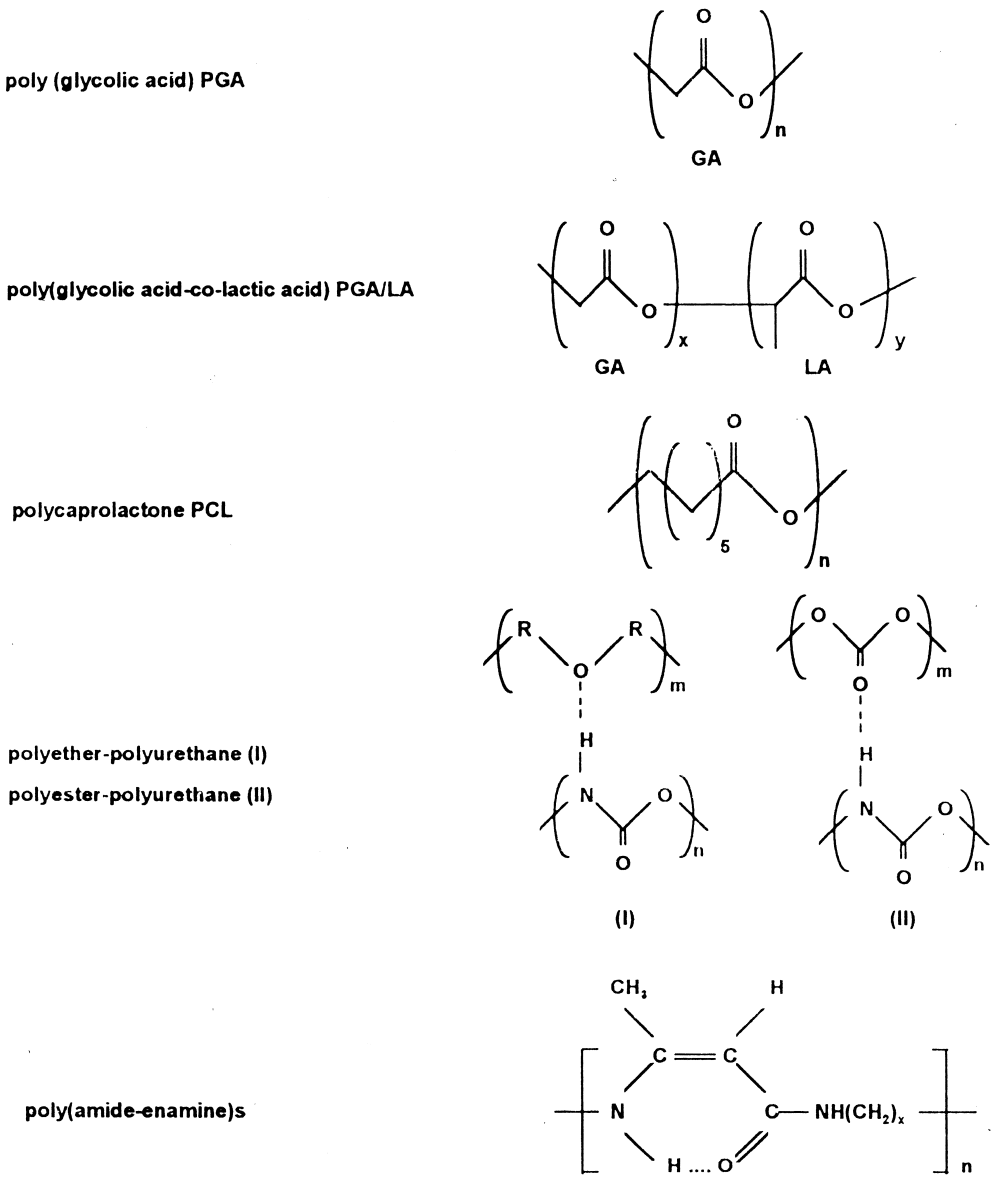


Fig. 3. Polymers with hydrolyzable backbone.

3.1. *Polyesters*

Almost the only high molecular weight compounds shown to be biodegradable are the aliphatic polyesters. The reason for this is the extremely hydrolyzable backbone found in these polyesters. It was found that polyesters derived from diacids of medium sized monomers (C_6–C_{12}) are more readily degraded by fungi (*Aspergillus niger* and *Aspergillus flavus*), than those derived from longer or shorter monomers.[76,77] In order for a synthetic polymer to be biodegradable by enzyme catalysts, the polymer chain must be able to fit into the enzyme's active site. This is one reason why flexible aliphatic polyesters are degradable and the rigid aromatic polyesters are not.[78–80]

Poly(glycolic acid) (PGA) is the simplest linear, aliphatic polyester. PGA[81–84] and poly(glycolic acid-*co*-lactic acid) (PGA/PL) are used as degradable and absorbable sutures. Their great advantage is their degradability by simple hydrolysis of the ester backbone in aqueous environments such as body fluids. Furthermore, the degradation products are ultimately metabolized to carbon dioxide and water or are excreted via the kidney.

3.2. *Polycaprolactone*

Poly(ϵ-caprolactone) (PCL) has been thoroughly studied as a substrate for biodegradation[85–92] and as a matrix in controlled-release systems for drugs.[93–96] Its degradation *in vivo* is much slower than that of poly (α-hydroxy acid)s.[93] Thus, it is most suitable for controlled-release devices with longer working lifetimes (1–2 years). PCL is generally prepared from the ring-opening polymerization of ϵ-caprolactone.[97] Tokiwa and Suzuki[98] have discussed the hydrolysis of PCL and biodegradation of PCL by fungi, and have shown that PCL can be degraded enzymatically.

Blends of PCL and polyesters prepared from alkanediols and alkane dicarboxylic acids with natural substances such as tree bark have been moulded into shaped containers for horticultural seeding plantouts.[97] After three months of soil burial, the PCL containers were found to be embrittled, disintegrated, and biodegraded which suggests that the extracellular enzymes in the soil may cleave the polymer chain prior to the assimilation of the polymer by microorganisms.

Polyesters derived from alkanediols and alkane dicarboxylic acids are readily degraded by biological systems[99–102] but their applications have been limited because of their relatively low molecular weights and poor physical strengths.

3.3. *Polyamides*

Although polyamides contain the same amide linkage that is found in polypeptides, their rate of biodegradation is so low that often they are reported to be nondegradable. However, the degradation by enzymes and microorganisms for low molecular weight oligomers has been reported.[103–107] Even aramid fibre was reported to be attacked by Aspergillus fungi.[108] The introduction of substituents such as benzyl, hydroxy and methyl greatly improve the biodegradation.

The higher crystallinity of polyamides due to strong interchain interactions (as compared with the more flexible polyesters with analogous structures), is behind the observed lower rates of biodegradation. Copolymers with both amide and ester groups are generally found to

be readily degraded.[109–114] As expected, the rate of degradation increases with increasing ester content.

Natural proteins seldom contain repeating units. As a result, there is less tendency for them to pack into highly ordered morphologies. Therefore, they are generally accessible to enzyme attack. On the other hand, synthetic polyamides have short and regular repeating units. Their higher symmetries and strong interchain hydrogen bonding result in highly ordered crystalline morphologies, which, in turn, limits the accessibility to enzyme attack. Poly(amide-ester)s and poly(amide-urethane)s with long repeating chains have been found to be degraded at rates somewhat in between those of proteins and synthetic polyamides.[109,114]

3.4. *Polyurethanes and polyureas*

Polyurethanes can be considered to have both the structural characteristics of polyesters and polyamides, whereas polyureas might be viewed as poly(diamide)s. Their susceptibility to biodegradation can be expected to be similar to that of polyesters and polyamides, with differences in rates. In general the biodegradability of polyurethanes was shown to be dependent on whether the prepolymer is a polyester or a polyether.[115] The polyether-based polyurethanes are resistant to biodegradation whereas the polyester polyurethanes are readily attacked. Many microorganisms (*Aspergillus niger*, *Aspergillus funeigatus*, *Fusarium solanii*, *Cryplococcus lacirentii*, etc.) and enzymes (papain, subtilisin, etc.) are effective in degrading polyurethanes. A series of polyurethanes derived from poly(caprolactone diol)s of various molecular weights, and aliphatic or aromatic diisocyanates were treated with various organisms. It was found that the degradation rate increases with increasing polyester segment length. It was also observed that polyurethanes derived from aliphatic diisocyanates are degraded faster than those derived from aromatic diisocyanates.[116]

3.5. *Polyanhydrides*

Polyanhydrides are a group of polymers with two sites in the repeating unit susceptible to hydrolysis. These are interesting materials due to their good biocompatibilities.[117] These are fibre-forming polymers that are very susceptible to hydrolysis.[118] Langer *et al*.[119] synthesized aliphatic–aromatic polyanhydrides for slow release formulations. The bioerodible polymers, especially polyanhydrides, are useful materials for drug delivery. The degradation rates can be altered with changes in the polymer backbone. Aliphatic polyanhydrides degrade within a few days while aromatic polyanhydrides can degrade slowly over a period of several years.[120] Recently, a new synthetic route for producing linear poly(adipic anhydride)s by use of ketene gas has been presented.[121] This synthetic route has the advantage of avoiding formation of acetic acid, which can drive the reaction backwards. Polyanhydrides are useful in biomedical applications due to their fibre-forming properties. An increase in the aliphatic chain length between the acid groups not only increases their molecular weight but also notably improves their hydrolytic stability.[122,123]

3.6. *Poly(amide-enamine)s*

The erosion of hydrophilic biodegradable polymer matrix systems such as PGA and poly(lactic acid) PLA or their copolymers generally proceeds in a homogeneous manner with

a progressive loosening or swelling of the matrix.[124] This changes the properties and release rate of the device. It is more desirable to have a matrix that can erode heterogeneously, e.g. by surface erosion, so that a near-zero-order release rate might be obtained if the diffusion release is small.[125] A hydrophobic polymer, yet one degradable by hydrolysis, is ideal for this purpose. Polyanhydrides show promising properties. Poly(amide-enamine)s have also been designed and synthesized for this purpose and have been found to be susceptible to hydrolysis and biodegradation, both by fungi and enzymes.[126]

4. POLYMERS WITH CARBON BACKBONES

Vinyl polymers, with few exceptions, are generally not susceptible to hydrolysis. Their biodegradation, if it occurs at all, requires an oxidation process, and most of the biodegradable vinyl polymers contain an easily oxidisable functional group. Approaches to improve the biodegradability of vinyl polymers often include the addition of catalysts to promote their oxidation or photooxidation, or both. The incorporation of photosensitive groups, e.g. ketones, into these polymers has also been attempted.

4.1. *Poly(vinyl alcohol) and poly(vinyl acetate)*

Poly(vinyl alcohol) (PVA) is the most readily biodegradable of vinyl polymers. It is readily degraded in waste-water-activated sludges.[127] The microbial degradation of PVA has been studied, as well as its enzymatic degradation by secondary alcohol peroxidases isolated from soil bacteria of the *Pseudomonas* strain.[128–131] It was concluded that the initial biodegradation step involves the enzymatic oxidation of the secondary alcohol groups in PVA to ketone groups. Hydrolysis of the ketone groups results in chain cleavage. Other bacterial strains, such as *Flavobacterium*[131] and *Acinetiobacter*[132] were also effective in degrading PVA.

The controlled chemical oxidation of PVA was carried out to yield poly(enol-ketone) (PEK), which has a similar structure to the intermediate formed as PVA is biodegraded.[133]

PEK was found to be much more susceptible to hydrolysis and biodegradation than PVA.[134,135] Since it is the polymeric form of acetoacetone, it undergoes chemical processes similar to those of acetoacetone, e.g. it forms metal chelates. Its water solubility, reactivity, and biodegradability make it a potentially useful material in biomedical, agricultural, and water treatment areas, e.g. as a flocculant, metal-ion remover, and excipient for controlled-release systems. By using dyes as models, it was found that PEK and PCL blends are excellent controlled-release matrix materials. The water-soluble PEK acts as excipient, whereas the hydrophobic and water-insoluble PCL acts as a barrier, keeping the device dimensions intact during the release period.[136]

Poly(vinyl acetate) (PVAC) reportedly undergoes biodegradation more slowly.[85,137,138] Copolymers of ethylene and vinyl acetate were susceptible to slow degradation in soil-burial tests.[139] The weight loss in a 120-day period increased with increasing acetate content. Because PVA is obtained from the hydrolysis of PVAC, which can be controlled easily in terms of the extent of hydrolysis and the sequence of PVAC and PVA, a controlled hydrolysis of PVAC followed by controlled oxidation should provide degradation materials having a wide range of properties and degradability.

PVA can form complexes with a number of compounds and has been used in the detoxification of organisms.[140] When it is used in a low-molecular weight form, i.e. below 15 000,

it can be eliminated from organisms by glomerular filtration. PVA has also been used as a polymer carrier for pesticides and herbicides.[141,142]

4.2. *Polyacrylates*

Poly(alkyl acrylate)s and polycyanoacrylates generally resist biodegradation.[85] Weight loss in soil-burial tests has been reported for copolymers of ethylene and propylene with acrylic acid, acrylonitrile, and acrylamide.[143] Poly(alkyl 2-cyanoacrylate)s, rapidly polymerizable systems adhering to moist surfaces, have been examined in biomedical applications.[144–149] Poly(methyl-2-cyanoacrylate) is the most degradable among the alkyl esters; degradability decreases as alkyl size increases. Poly(isobutyl-2-cyanoacrylate) nanoparticles have been degraded in two enzyme-free media at pH 7 and 12 in the presence of rat liver microsomes. It was found that the formaldehyde-producing degradation route is less efficient, and the ester hydrolysis is catalyzed by enzymes. The release rate of adsorbed actinomycin from nanoparticles correlated well with the degradation of the poly(isobutyl-2-cyanoacrylate).

Poly(2-hydroxyethyl methacrylate) is generally cross-linked with a small amount of ethylene dimethacrylate. It swells in water to form a hydrogel and has been widely used in biomedical areas because of its good biocompatibility.[150–154] Although earlier papers reported its inertness under *in vivo* and *in vitro* conditions,[152,155] more recent work has indicated that it slowly hydrolyzes *in vitro*.[156]

The need for a spacer molecule between a bound drug and the carrier polymer in order to achieve effective cleavage in some biological systems has long been known.[157] For example, androgen has been bound covalently to a copolymer of methacrylic and acrylic acid with and without a spacer.[158,159] These compounds were then injected subcutaneously into castrated rats and the amount of androgen in the lavatory muscle, the prostate gland, and the sperm duct was determined. To increase the biodegradability of poly(*N*-(2-hydroxypropyl) methyl acrylamide), biodegradable segments, e.g. peptides, have been incorporated into the polymer chains.

5. FACTORS AFFECTING BIODEGRADATION

5.1. *Effect of polymer structure*

Natural macromolecules, e.g. protein, cellulose, and starch are generally degraded in biological systems by hydrolysis followed by oxidation. It is not surprising, then, that most of the reported synthetic biodegradable polymers contain hydrolyzable linkages along the polymer chain; for example, amide enamine, ester, urea, and urethane linkages are susceptible to biodegradation by microorganisms and hydrolytic enzymes. Since many proteolytic enzymes specifically catalyze the hydrolysis of peptide linkages adjacent to substituents in proteins, substituted polymers containing substituents such as benzyl, hydroxy, carboxy, methyl, and phenyl groups have been prepared in the hope that an introduction of these substituents might increase biodegradability.[109]

Among benzylated polymers, mixed results have been obtained for polyamides. The achiral poly(hexamethylene-α-benzylmalonamide) is hydrolyzed readily by chymotrypsin, an enzyme known to catalyze the hydrolysis of peptide linkages adjacent to the benzyl group of the phenylalanine residues in proteins specifically. On the other hand, poly(alkylene D,L-α-benzyladipamide)s have very low biodegradabilities.

Apparently, the chiral specificity of enzymes are maintained here. In an investigation designed to study the effects of stereochemistry on the biodegradation of polymers, monomeric and polymeric ester-ureas were synthesized from D-, L-, and D,L-phenylalanines.[160] When subjected to enzyme-catalyzed degradation, the pure L-isomer was degraded much faster than the D,L-isomers. Chymotrypsin was also effective in degrading benzyl-substituted poly(ester-urea)s derived from phenylalanine, but not in degrading the unsubstituted poly(ester-urea)s derived from glycine. This agreed with the well-known substituent specificity of chymotrypsin.

Since most enzyme-catalyzed reactions occur in aqueous media, the hydrophilic–hydrophobic character of synthetic polymers greatly affects their biodegradabilities. A polymer containing both hydrophobic and hydrophilic segments seems to have a higher biodegradability than those polymers containing either hydrophobic or hydrophilic structures only. A series of poly(alkylene tartrate)s was found to be readily assimilated by *Aspergillus niger*. However, the polymers derived from C_6 and C_8 alkane diols were more degradable than the more hydrophilic polymers derived from C_2 and C_4 alkane diols or the more hydrophobic polymers derived from the C_{10} and C_{12} alkane diols. Among the degradable poly(α-amino acid-*co*-ϵ-caproic acid)s, the hydrophilic copolyamide derived from serine was more susceptible than those containing only hydrophobic segments.[161]

In order for a synthetic polymer to be degradable by enzyme catalysis, the polymer chain must be flexible enough to fit into the active site of the enzyme. This most likely accounts for the fact that, whereas the flexible aliphatic polyesters are readily degraded by biological systems, the more rigid aromatic poly(ethylene terephthalate) is generally considered to be bioinert.[85,101]

5.2. *Effect of polymer morphology*

One of the principal differences between proteins and synthetic polymers is that proteins do not have equivalent repeating units along the polypeptide chains. This irregularity results in protein chains being less likely to crystallize. It is quite probable that this property contributes to the ready biodegradability of proteins. Synthetic polymers, on the other hand, generally have short repeating units, and this regularity enhances crystallization, making the hydrolyzable groups inaccessible to enzymes. It was reasoned that synthetic polymers with long repeating units would be less likely to crystallize and thus might be biodegradable; indeed, a series of poly(amide-urethane)s were found to be readily degraded by subtilisin.[109]

Selective chemical degradation of semicrystalline polymer samples shows certain characteristic changes.[162–170] During degradation, the crystallinity of the sample increases rapidly at first, then levels off to a much slower rate as the crystallinity approaches 100%. This is attributed to the eventual disappearance of the amorphous portions of the sample. The effect of morphology on the microbial and enzymatic degradation of PCL, a known biodegradable polymer with a number of potential applications, has been studied.[86–89] Scanning electron microscopy (SEM) has shown that the degradation of a partially crystalline polycaprolactone film by filamentous fungi proceeds in a selective manner, with the amorphous regions being degraded prior to the degradation of the crystalline region. The microorganisms produce extracellular enzymes responsible for the selective degradation. This selectivity can be attributed to the less-ordered packing of amorphous regions, which permits easier access for the enzyme to the polymer chains. The size, shape and number of the crystallites all have a

pronounced effect on the chain mobility of the amorphous regions and thus affect the rate of the degradation. This has been demonstrated by studying the effects of changing orientation via stretching on the degradation.[87–89]

Biodegradation proceeds differently from chemical degradation. Studies on the degradation by solutions of 40% aqueous methylamine have shown a difference in morphology and molecular weight changes and in the ability of the degrading agents to diffuse into the substrate. Also, it was found that the differences in degradation rates between amorphous and crystalline regions are not same. The enzyme is able to degrade the crystalline regions faster than can methylamine. Quantitative GPC (gel permeation chromatography) analysis shows that methylamines degrade the crystalline regions, forming single and double transverse length products. The enzyme system, on other hand, shows no intermediate molecular weight material and much smaller weight shift with degradation. This indicates that although degradation is selective, the crystalline portions are degraded shortly after the chain ends are made available to the exoenzyme. The lateral size of the crystallites has a strong effect on the rate of degradation because the edge of the crystal is where degradation of the crystalline material takes place, due to the crystal packing. A smaller lateral crystallite size yields a higher crystallite edge surface in the bulk polymer. Prior to the saturation of the enzyme active sites, the rate is dependent on available substrate; therefore, a smaller lateral crystallite size results in a higher rate of degradation. The degradation rate of a PCL film is zero order with respect to the total polymer, but is not zero order with respect to the concentrations of the crystallite edge material. The drawing of PCL films causes an increase in the rate of degradation, whereas annealing of the PCL causes a decrease in the rate of degradation. This is probably due to opposite changes in lateral crystallite sizes.

In vitro chemical and enzymatic degradations of polymers, especially polyesters, were analyzed with respect to chemical composition and physical properties. It was found quite often that the composition of a copolymer giving the lowest melting point is most susceptible to degradation.[171] The lowest packing order, as expected, corresponds with the fastest degradation rate.

5.3. *Effect of radiation and chemical treatments*

Photolysis with UV light and the γ-ray irradiation of polymers generate radicals and/or ions that often lead to cleavage and crosslinking. Oxidation also occurs, complicating the situation, since exposure to light is seldom in the absence of oxygen. Generally this changes the material's susceptibility to biodegradation. Initially, one expects the observed rate of degradation to increase until most of the fragmented polymer is consumed and a slower rate of degradation should follow for the crosslinked portion of the polymer. A study of the effects of UV irradiation on hydrolyzable polymers confirmed this.[172] Similarly, photooxidation of polyalkenes promotes (slightly in most cases) the biodegradation.[173,174] The formation of carbonyl and ester groups is responsible for this change.

Processes have been developed to prepare copolymers of alkenes containing carbonyl groups so they will be more susceptible to photolytic cleavage prior to degradation. The problem with this approach is that negligible degradation was observed over a two year period for the buried specimens. Unless a prephotolysis arrangement can be made, the problem of plastic waste disposal remains serious, as it is undesirable to have open disposal, even with constant sunlight exposure.

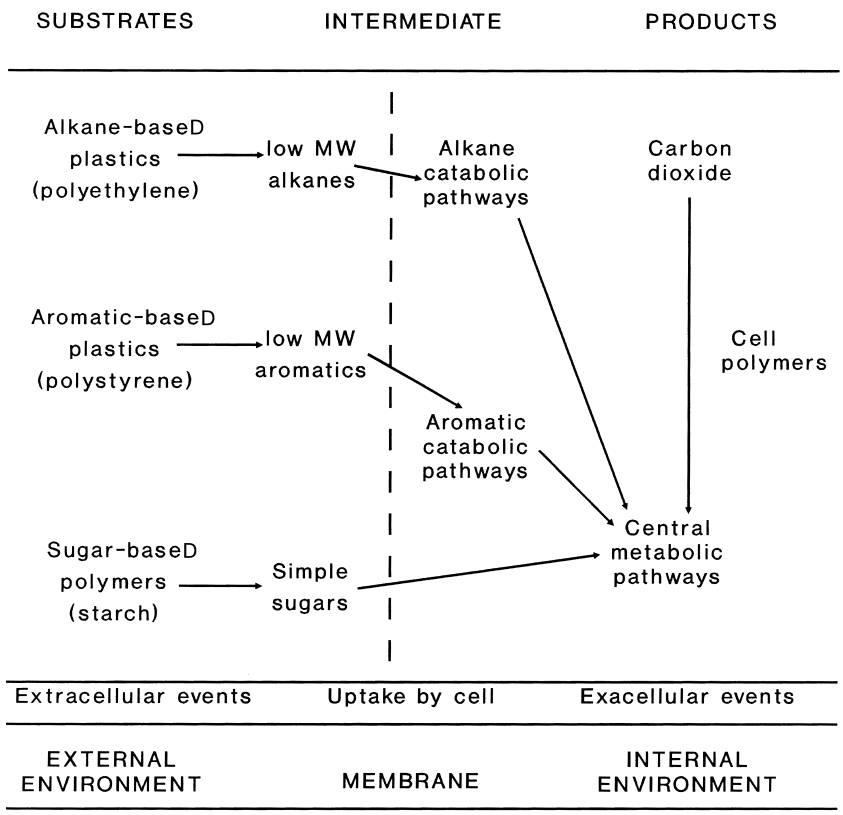


Fig. 4. Pathways for polymer biodegradation.

As expected, γ-ray irradiation greatly affects the rate of *in vitro* degradation of polyesters.[175,176] For polyglycolide and poly(glycolide-*co*-lactide), the pH of the degradation solution decreased as the process proceeded. The change-time curves exhibit sigmoidal shapes and consist of three stages: early, accelerated, and later; the lengths of these three regions were a function of γ-ray irradiation. Increasing radiation dosage shortens the time of the early stage. The appearance of the drastic pH changes coincides with loss of tensile breaking strength. Similar effects via enzymatic and microbial degradation remain to be demonstrated.

5.4. *Effect of molecular weight*

There have been many studies on the effects of molecular weight on biodegradation processes. Most of the observed differences can be attributed to the limit of detecting the changes during degradation, or, even more often, the differences in morphology and hydrophilicity–hydrophobicity of polymer samples of varying molecular weight. Microorganisms produce both exoenzymes [degrading polymers from terminal groups (inwards)] and endoenzymes (degrading polymers randomly along the chain). One might expect a large molecular effect on the rate of degradation in the ease of exoenzymes and a relatively small

molecular weight effect in the case of endoenzymes. Plastics remain relatively immune to microbial attack as long as their molecular weight remains high. Many plastics, such as PE, PP and PS do not support microbial growth. Low molecular weight hydrocarbons, however, can be degraded by microbes. They are taken in by microbial cells, 'activated' by attachment to coenzyme-A, and converted to cellular metabolites within the microbial cell, as shown in Fig. 4. However, these processes do not function well (if at all) in an extracellular environment, and the plastic molecules are too large to enter the cell. This problem does not arise with natural molecules, such as starch and cellulose, because conversions to low molecular weight components by enzyme reactions occur outside the microbial cell. Photodegradation or chemical degradation may decrease molecular weight to the point that microbial attack can proceed, however.

The upper limits of molecular weight, beyond which uptake and intracellular degradation do not occur, have not been established for all alkane-derived materials. Very slow degradation of paraffins, PE glycols, and linear alkyl benzene sulphonates occurs when the length of the polymer chain exceeds 24–30 carbon atoms.[177–179] It could be concluded from these amply documented results that alkane-based plastics with molecular weights exceeding 400–500 daltons (i.e. greater than 30 carbon atoms) must be degraded into smaller molecules by photodegradation, chemical or other biological means before biodegradation. LDPE with a molecular weight average of $M_w = 150\,000$ contains about 11 000 carbon atoms. Decreasing molecules of this size to biologically acceptable dimensions requires extensive destruction of the PE matrix. This destruction can be partly accomplished in blends of PE and biodegradable natural polymers by the action of organisms, such as arthropods, millipedes, crickets, and snails.

Insertion of carbon monoxide into the chain permits chain scission by a Norrish-type reaction in a photochemical process.[180] It was found that E/CO polymers with 2.5% CO linkages lost about 98% of their original elongation after 40 h of sunlamp exposure. However, after 650 h of exposure, the samples that originally had $M_w = 618\,700$ and $M_n = 45\,000$ had photolytic products with $M_w = 15\,000$ and $M_n = 7300$.

6. MODE OF BIODEGRADATION

The biological environment, i.e. the biological surroundings in which polymers are present, includes the biological agents responsible for the deterioration of polymeric substances. Biological agents such as bacteria, fungi and their enzymes consume a substance as a food source so that its original form disappears. Under appropriate conditions of moisture, temperature, and oxygen availability, biodegradation is a relatively rapid process.

6.1. *Microorganisms*

Two types of microorganisms are of particular interest in the biodegradation of natural and synthetic polymers: bacteria and fungi.

6.1.1. *Fungi*

Eumycetes, or true fungi, are microorganisms of particular importance in causing the degradation of materials. Fungi are nucleated, spore-forming, nonchlorophyllous organisms,

which reproduce both sexually and asexually; most of them possess filamentous, somatic structures, and cell walls of chitin and/or cellulose. More than 80 000 species are known.

True fungi are present everywhere. Their importance as deteriorative agents is a result of the production of enzymes which break down nonliving substrates in order to supply nutrient materials present in polymer compositions. Certain environmental conditions are essential for optimum growth and degradative activity. These include an optimal ambient temperature, the presence of nutrient materials, and high humidity.

The group of test fungi that evolved for assay purposes in the field of natural polymers and that were further selected for their utility in assay procedures on synthetic polymers are taxonomically a very heterogeneous group, exhibiting no marked taxonomic similarities among them (for example based on morphology). Many of them were selected primarily because their reproduction spores are produced asexually and the variation associated with spores resulting from the fusion of sexual element is minimized. The test organisms cited are also, for the most part, the selected organisms from a large number of isolations which have proved their capability for yielding reproducible results repetitively, over long periods of time, under laboratory conditions, and in synthetic or highly controlled and specific culture media.

The most acceptable organisms are characterized by strain or culture collection number. The strain of *Aspergillus niger* is identified by the ATCC Number 9642 or the Quartermaster (QM) Number or Mycological Services No. 386.

6.1.2. *Bacteria*

Schizomycetes, a bacteria, have played an undetermined role in relation to fungi in polymer deterioration. Bacteria can be single-cell rods, cocci, or spirilla; others are chain-like or filamentous. Bacteria can either be aerobic or anaerobic; in contrast, fungi are necessarily aerobic. Some bacteria are motile; bacteria are predominantly nonchlorophyllous. Their degradative action is also chiefly a result of enzyme production and resultant breakdown of the nonliving substrate in order to obtain nutrient materials.

Bacteria present in soil are important agents for material degradation. Particularly affected are cellulosic plant life, wood products, and textiles subject to cellulytic degradation.

6.2. *Enzymes*

Enzymes are essentially biological catalysts, with the same action as chemical catalysts. By lowering the activation energy they can induce an increase in reaction rates in an environment otherwise unfavourable for chemical reactions, e.g. water at pH 7 and 30°C. In the presence of enzymes, a rise in reaction rate of 10^8–10^{20} can often be observed. The vast majority of enzymes are proteins having a polypeptide chain with a complex three-dimensional structure. Enzyme activity is closely related to conformational structure.

The three-dimensional structure of enzymes with folds and pockets creates certain regions on the surface with characteristic primary structures (i.e. specific amino acid sequences) which form an active site. At the active site the interaction between the enzyme and substrate takes place leading to a chemical reaction, giving a particular product.

For optimal activity certain enzymes must associate with cofactors which can be metal ions, e.g. sodium, potassium, magnesium, calcium or zinc. Organic cofactors are also called

coenzymes and they can vary in structure, some are derived from different B-vitamins (thiamine, biotin, etc.) while others are important compounds in metabolic cycles such as nicotinamide adenine dinucleotide (NAD^+), nicotinamide adenine dinucleotide phosphate ($NADP^+$), flavin adenine dinucleotide (FAD^+), adenosine triphosphate (ATP), etc. An enzyme plus a cofactor is called a holoenzyme while an enzyme lacking a cofactor is called an apoenzyme.

All enzymes, except those few retaining historically important trivial names (trypsin, pepsin, etc.), are named according to rules adopted by the International Enzyme Commission. The names give the nature of the chemical reaction catalyzed and also describe the substrate. All new enzymes end with the suffix -ase, but shorter names are often used as some enzyme names become very long, e.g. hexokinase for ATP:hexose-phosphotransferase.

For enzymes with absolute specificities the 'key-and-lock' theory which implies an unchangeable rigid conformation, is a plausible model. The initial contact between an enzyme and substrate forms an optimal orientation at the active site giving good possibilities for maximum bonding (enzyme–substrate), often the cofactor induces these changes when binding to the enzyme.

6.2.1. *Physical factors affecting the activity of enzymes*

All enzymes are adjusted to a specific environment in which their activity and three-dimensional structure are optimal for a specific purpose. For human enzymes or enzymes isolated from human cells, this environment is a water solution at pH 6–8, an ion strength of 0.15 molar (as is normal physiological saline at 0.9% NaCl) and a temperature of 35–40°C. An extremely small change one of these parameters may render the enzyme totally inactive and sometimes can even destroy it irreversibly. Other solvents than water, especially organic solvents, are also lethal to many enzymes but, on other hand, there are enzymes that are active in extreme environments, e.g. in hot water springs or salty environments.

6.2.2. *Enzyme mechanisms*

Different enzymes have different actions, some enzyme change the substrate through a free radical mechanism while others follow alternative chemical routes. Typical examples are biological oxidation and biological hydrolysis.

6.2.2.1. *Biological oxidation* – Several enzymes can react directly with oxygen, the classical example being cytochromoxidase which is active in the respiratory chain. Oxygen has a special role in the metabolism of aerobic organisms. In many cases oxygen is directly incorporated into the substrate. The enzyme can be hydroxylases (Fig. 5, eq. 1) or oxygenases (Fig. 5, eq. 2).

Hydroxylases are sometimes called monooxynases and catalyze the insertion of a single atom of oxygen in the substrate A as part of a hydroxyl group. The monooxynases require a second reduced substrate BH_2 which simultaneously undergoes oxidation (i.e. dehydrogenation). Usually this second substrate is NADH (NADPH).

Oxygenases, also called dioxygenases, catalyze the insertion of a whole oxygen molecule into the substrate, sometimes the product is a dihydroxy derivative but more often the oxygen atoms are incorporated as a part of a carbonyl (–CO) or a carboxyl (–COO–) grouping.

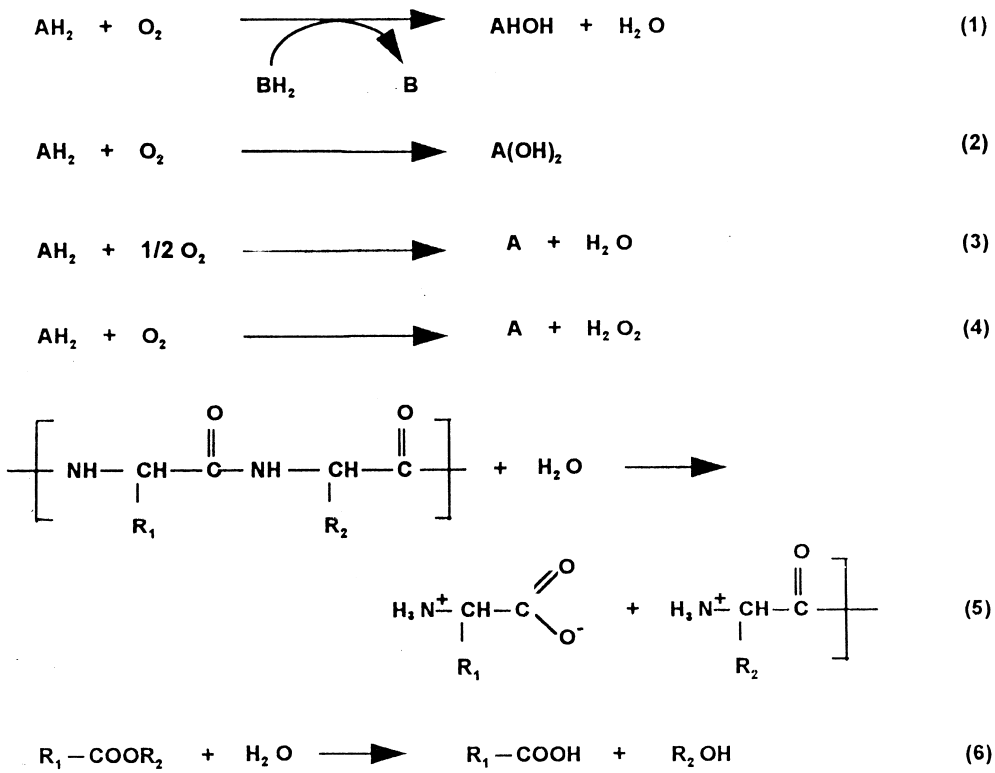


Fig. 5. Biological oxidation and hydrolysis by enzymes.

Yet another type of biological oxidation exists, namely the process where the oxygen molecule is not actually incorporated into the substrate, but rather it functions as a hydrogen acceptor (i.e. electron acceptor). Enzymes of this type are called oxidases and one type produces H_2O (Fig. 5, eq. 3) while another produces H_2O_2 (Fig. 5, eq 4).

One example of an oxygenase enzyme is that capable of catalyzing the splitting of aromatic structures producing (–C=O) groups instead of the (–HC=CH–) group.

6.2.2.2. *Biological hydrolysis* – Several different hydrolysis reactions occur in biological organisms. Proteolytic enzymes (proteases) catalyze the hydrolysis of peptide bonds (Fig. 5, eq. 5) and also the related hydrolysis of an ester bond (Fig. 5, eq. 6).

7. TEST METHODS AND STANDARDS FOR BIODEGRADABLE POLYMERS

The extent of the biodegradation of degradable plastics is undergoing considerable re-examination. Marine areas, soil, sewage, and composts represent complex biological environments. A large number of microorganisms from different species and genera are present in these environments. These microorganisms display a broad range of polymer-degrading

abilities ranging from the complete degradation of a polymer in one environment to the negligible degradation of the same polymer in another environment. Consequently, for a biodegradable polymer to be used for a certain application, the polymer should necessarily degrade in the environment of end usage and not necessarily degrade in other environments. For example, biodegradable agricultural mulch films should degrade when they are in contact with soil microorganisms but not necessarily degrade in a marine environment. Therefore, a good method for the evaluation of biodegradability should consider the end usage of the polymer and the environment of end usage. American Standard Testing Methods (ASTM) and the Organization for Economic Cooperation and Development (OECD) have proposed several test methods.

The use of decision trees is gaining popularity in following a programme of testing in a logical fashion to rule out the generation of unnecessary and irrelevant data. An example decision tree[181] is presented in Fig. 6. At any decision point the assessor is only faced with two alternative routes to follow which will be based upon the prior acquisition and interpretation of test data.

The first decision requires data on the concentration released and the compartment of the environment to which the product is released. If there is no likelihood that the polymer will ever come into contact with the biosphere then it is senseless carrying out a biodegradability assessment. This is, of course, a hypothetical situation. The placement of biodegradable polymers into the biosphere is highly likely and one or more routes of exposure must be defined. From this, a suitable methodology can be chosen for the first tier of testing.

If the polymer 'passes' this first tier then no further testing is necessary. 'Failing' the test triggers the decision to proceed to the next tier. Decisions are thus made in a stepwise fashion until the hypothesis that the polymer is biodegradable has been proved or not.

7.1. *Modified Sturm test*

The modified Sturm test seems to be the preferred technique for polymeric materials. It has been specified by the Italian authorities for assessing biodegradable polymers and is currently being evaluated by the Biodegradable Plastics Group of the International Biodeterioration Research Group.[182] The test operates under aerobic conditions, the test substance provides the sole source of carbon, it is exposed to a low level of inoculum and a nonspecific method of analysis is used to follow the course of biodegradation. The test is run for 28 days without any acclimatization period.

The process of the Sturm method is as follows. To a chemically defined mineral nutrient solution free of organic carbon, the test substance is added at two concentrations (10 and 20 mg l^{-1}). An inoculum of sewage microorganisms is added ($1–20 \times 10^6$ ml^{-1}) to the solution. The test system with suitable controls are incubated at ambient temperatures with stirring for 28 days. The CO_2 evolved is trapped in alkali and measured as carbonate by either titration or with the use of a carbon analyzer.

After analysis of the data with respect to suitable blank controls the total amount of CO_2 produced by the polymer over the test period is determined and calculated as that percentage of the total CO_2 which the polymer could have theoretically produced based upon its total carbon content. Because a proportion of carbon will be incorporated into biomass, the total CO_2 and hence calculated biodegradation levels can never reach 100%. With this in mind, more realistic levels have been recommended.

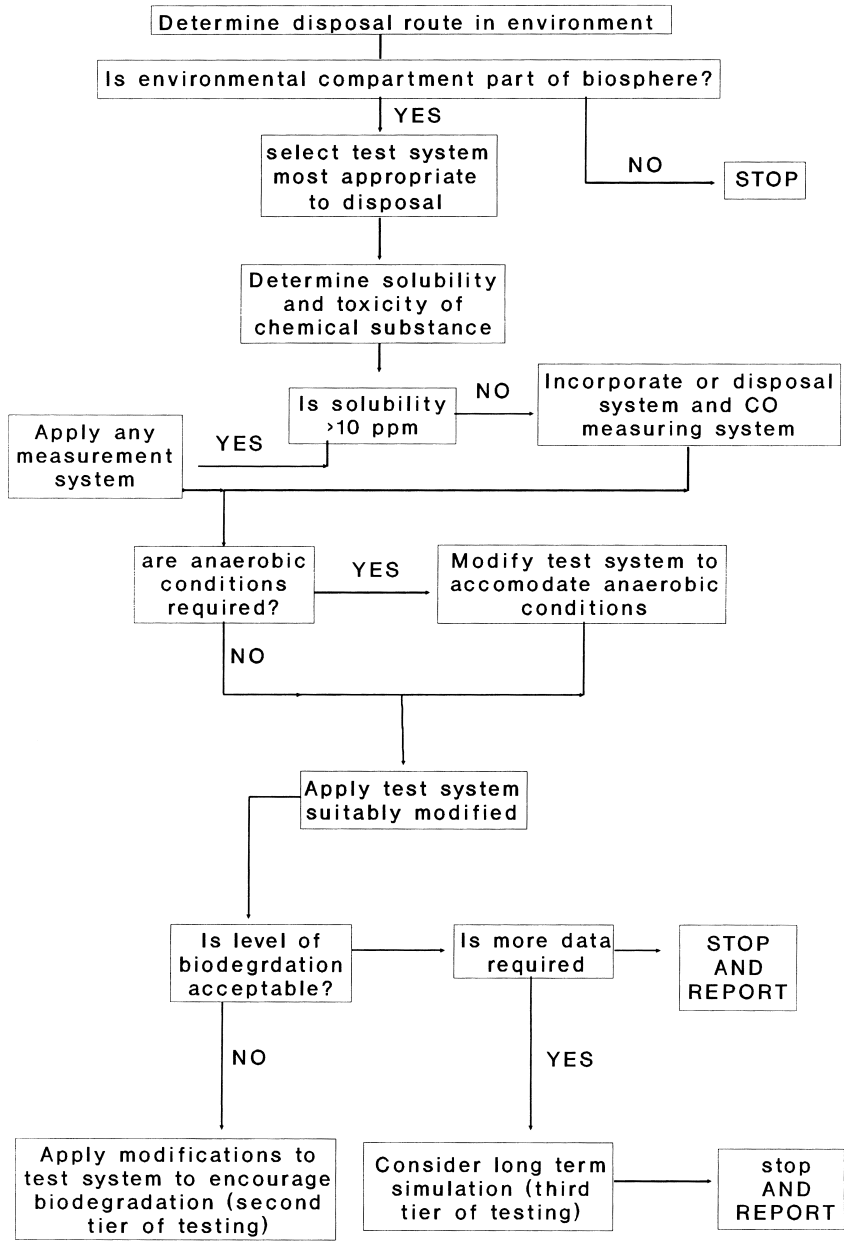


Fig. 6. Decision tree for evaluating biodegradability.

For a chemical substance to be regarded as readily biodegradable, it should produce greater than 60% of its theoretical total within 28 days. This level should be reached within 10 days of biodegradation reaching 10%.

The reproducibility of the test is quoted in the guidelines[183] as being + 5% but this is based

upon work with soluble materials. For insoluble materials, the form in which they are presented to the test system and the efficiency of their dispersion will affect the reproducibility. The use of a powder or teased fibres will give the best surface to volume ratio but care should be taken to ensure that clumping or adsorption to the walls of the test vessel is avoided.

An alternative approach is to determine the biochemical oxygen demand of the polymer over 28 days (BOD_{28}). This test method is probably the most stringent of the aqueous screening tests because of the low levels of inoculum used (of the order of 10^2 microorganisms per ml) and limited amount of test substance which can be added (normally between 2 and 4 mg l^{-1}). The calculation in arriving at the correct application level is based on the theoretical oxygen demand (determined by calculation) or chemical oxygen demand (determined experimentally) of the test substance being not more than one half of the maximum dissolved oxygen level in the water at the temperature of the test. This is determined as follows.

Taking glucose as an example, the theoretical or chemical oxygen demand is calculated as 1.07 mg O_2 per mg glucose and the concentration of dissolved O_2 in water at 20°C is about 9 mg l^{-1}. To ensure that 50% of the dissolved O_2 remains at the end of the test period the total oxygen demand must not exceed 4.5 mg l^{-1}, thus the maximum concentration of glucose used should be 4.2 mg l^{-1}.

The theoretical oxygen demand (TOD) of a chemical is TOD = 16[2c + h/2-0] molecular weight which requires a knowledge of the formula of the test substance. This may be difficult to determine accurately for polymeric materials and therefore a chemical oxygen demand will need to be carried out.

7.2. *Closed bottle test*

The closed bottle test method is as follows. A predetermined amount of the test substance is added to a chemically defined mineral salt solution. The solution is inoculated with sewage microorganisms and then dispersed into closed bottles. The bottles are incubated in the dark at 20 ± 1°C and periodically assessed for their dissolved oxygen contents. The oxygen demand is calculated and compared with theoretical or chemical oxygen demand of the test substance. Polymers have to be prepared as finely divided powders and their continuous dispersion in the nutrient solution assured. This can only effectively be done using magnetic stirring and this may preclude the use of this test as one test substance requires that at least 25 bottles be used.

7.3. *Petri dish screen*

This test is used in USA (ASTM), German (DIN), French (AFNOR), Swiss (SN) and international (ISO) standards (Table 1). The principle of this method involves facing the test material (2.5×2.5 cm^2) on the surface of mineral salts agar in a petri dish containing no additional carbon source. The test material and agar surface are sprayed or painted with a standardized mixed inoculum of known fungi or bacteria (Table 2). The petri dishes are sealed and incubated at a constant temperature between 21 and 28 days. The test material is then examined for the amount of growth on its surface and a rating given (Table 3). The more growth on the surface, the more likely it is that the material is intrinsically able to support growth and thus the greater the likelihood that it will fail in service.

Weight loss, mechanical or electrical tests can all be carried out on the test materials after

Table 1. Standards used for resistance testing of polymers

Title	Standard authority and member
Plastics: determination of behaviour under the action of fungus and bacteria	ISO 846
	NFX41-514
Basic environmental testing procedures for electronic equipment	BS2011 part 2.1
Test J, Mould growth	Test J
Resistance of plasticisers to attack by microorganisms	NFX41-513
Determination of the resistance of plastics to fungi and bacteria	DIN53 739
Standard practice for determination the resistance of synthetic polymeric material to fungi	ASTM G21-70
Standard practice for determining the resistance of synthetic polymeric materials to bacteria	ASTM G22-76
Standard practice for determining the resistance of synthetic polymeric materials to algae	ASTM G29-75

Notes: BS—British Standards Institute, DIN—German Standards Institute, ISO—International Standards Organisation, NFX—French Standards Institute.

Table 2. Test strains of fungi and bacteria used for resistance testing of plastics

Test strain	Culture collection number	Standard
Aspergillus niger	IMI 17454	BS 2011 Part 2.1
	IMI 45551	ISO 846
	IMI 91855	ASTM G21-70
Aspergillus terreus	IMI 45543	BS 2011 Part 2.1J
Aureobasidium pullulans	IMI45553	BS 2011 Part 2.1J
	IMI 145194	ISO 846
Chaetomium globosum	IMI 45550	ASTM G21-70
		ISO 846
Paecilomyces variotii	IMI 108007	BS 2011 Part 2.1J
		ISO 846
Penicillium funiculosum	IMI 114933	BS 2011 Part 2.1J
		ISO 846
Penicillium ochrochoron	IMI 61271i	BS 2011 Part 2.1J
Scopulariopsis brevicaulis	IMI 49528	BS 2011 Part 2.1J
Trichoderma viride	IMI 45553i	BS 2011 Part 2.1J
		ASTM G21-70
		ISO 846

exposure provided that the correct types of specimen (e.g. dumbbells) have been used in the test. The validity of this type of test and the use of visual assessment alone has been questioned by Seal and Pantke[184] for all plastics. They recommended that mechanical properties should give support to visual assessments. Such tests must be treated with caution when extrapolating the data.

7.4. *Environmental chamber method*

The environmental chamber employs high humidity (>90%) situations to encourage microbial (in particular fungal) growth. Strips or prefabricated components of the test materials

Table 3. Rating scheme based on visual assessment used by ISO 846 for assessing fungal resistance of plastics

Visual assessment	Rating	Evaluation
No growth apparent even under the microscope	0	The material is not a nutritive medium for microorganism
Growth invisible or hardly visible to the naked eye but clearly visible under the microscope	1	The material contains the nutritive substances
Slight growth covering less than 25% of the specimen surface	2	The material is not resistant to fungal attack and contains nutritive substances
Growth covering more than 25% of the specimen surface	3	As for rating 2

are hung in the chamber, sprayed with a standard mixed inoculum of known fungi (Table 2) in the absence of additional nutrients and then incubated for 28 to 56 days at a constant temperature. A visual assessment is subsequently made and a rating given based on the amount of growth on the material (Table 3). This test is particularly stringent and was designed to simulate the effects of high humidity conditions on electronic components and electrical equipment. The growth of fungi across a printed circuit board can result in a gross system failure in a computer system or in military equipment. Such a test is valuable in assessing how biodegradable polymers will perform under such conditions whilst in service.

7.5. *Soil burial test*

Tests based upon this methodology evaluate in-service soil contact exposure. The material is buried in soil beds prepared in the laboratory using standard sieved soil; often a commercial soil-based compost. The soil beds are normally conditioned for up to four weeks prior to use and may be supplemented with organic fertilizer to encourage an active microbial flora. The microbial activity is tested using a cotton textile strip which should lose 90% of its tensile strength within 10 days of exposure to the soil. Currently no other reference materials are recommended, although for plastic materials a standard alternative able to demonstrate the degradation capabilities of the microbial flora with respect to plastic should be sought. The soil beds containing the samples are incubated at a constant temperature for between 28 days and 12 months. The moisture content is normally set at 20–30%, although it is better calculated as a percentage (40–50%) of the soil's maximum water holding capacity. This then accounts for different soil structures and ensures that the soil does not become unduly wetted or is too dry for optimal microbial activity. Samples are removed for assessment of changes or a light microscopy and SEM examination to assess surface damage and to look for the presence and nature of microbial growth. Physical factors such as fragmentation and embrittlement can also be assessed in these tests. Finally, the samples can be used to 'bait' microorganisms involved in the degradation process. These microbes once isolated and characterized can be incorporated into the petri dish screen as alternatives or additions to the current list.

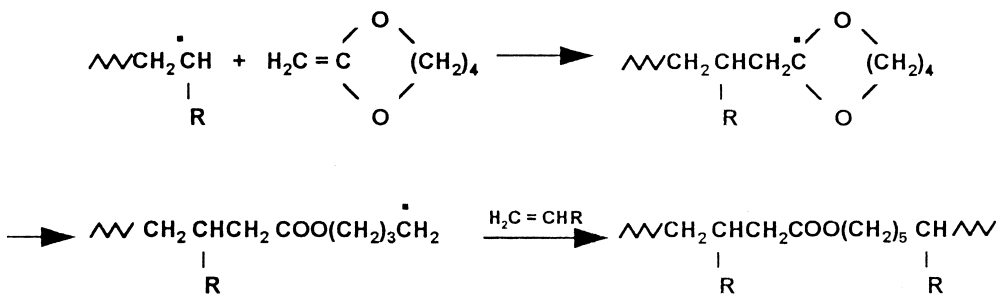


Fig. 7. Insertion of ester group into vinyl polymer.

8. POLYMER MODIFICATION TO FACILITATE BIODEGRADATION

Because oligomers and polymers with main chains containing only carbon–carbon bonds (except for those with large numbers of polar groups on the main chain such as poly(vinyl alcohol)) show little or no susceptibility to enzyme-catalyzed degradation reactions, especially at higher molecular weights, several approaches have been used to insert 'weak links' within or immediately attached to the backbones of such polymers.[185] These 'weak links' are designed to permit the controlled degradation of an initially high molecular weight, hydrophobic polymer into a lower molecular weight oligomer, which can then be utilized and consumed by microorganisms through biodegradation processes. Particular emphasis in this approach to create useful biodegradable polymers has been placed on two types of polymer modifications: namely the insertion of functional groups in the main chain, especially ester groups, which can be cleaved by chemical hydrolysis, and the insertion of functional groups in or on the main chain that can undergo photochemical chain-cleavage reactions, typically carbonyl groups.

An exceedingly clever method for inserting main chain ester groups into vinyl-type polymers (Fig. 7), including polystyrene and polyethylene, is to carry out a free radical copolymerization reaction on the appropriate vinyl monomer (e.g. styrene or ethylene) with a special monomer that undergoes free radical, ring opening reaction to generate a main chain ester group.[186,187] Methylene-substituted cyclic acetal and ortho-ester monomers can participate in such a reaction by free radical copolymerization according to the mechanism shown for one of these types of comonomers: 2-methyleneoxepane (R = H for polyethylene or R = C_6H_5 for polystyrene-based copolymers).

By using this procedure several different types of otherwise all-carbon chain polymers have been prepared and after hydrolysis to their lower molecular weight carboxy- and hydroxyl-terminated oligomers, the latter can be degraded by fungal attack.

The other principal approach, the preparation of a photodegradable copolymer, also utilizes a free radical copolymerization reaction. But in this case the comonomer is one which will create a ketone group either in the main chain or immediately attached to the main chain (Fig. 8). Both carbon monoxide and vinyl ketones will form such 'weak links', and both of these comonomers have been used effectively in small amounts to prepare useful copolymers with a variety of vinyl-type monomers (again, especially for polystyrene, R = C_6H_5, and polyethylene, R = H).[188,189]

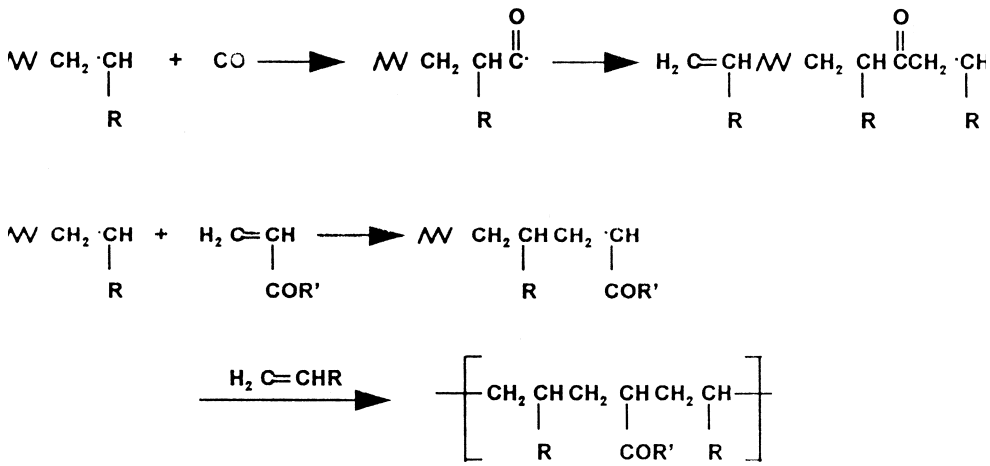


Fig. 8. Insertion of ketone group into vinyl polymer.

On irradiation with ultraviolet light, the activated ketone groups present can take part in two different types of free radical, bond-breaking reactions. In organic photochemistry, these two reactions are referred to as Norrish I and Norrish II reactions, and their mechanisms are shown in Fig. 9 for the degradation of copolymers of ethylene and carbon monoxide.[189,190]

The Norrish I reaction fragments the polymer to generate both carbonyl and alkyl radicals, and in the presence of oxygen, these reactive fragments form carboxylic acid-terminated and hydroxyl-terminated lower molecular weight chains. Further reactions of this type generate oligomers that can presumably interact with enzymes by the processes described above for the biodegradation of alkanes. The Norrish II reaction creates fragments with vinyl and

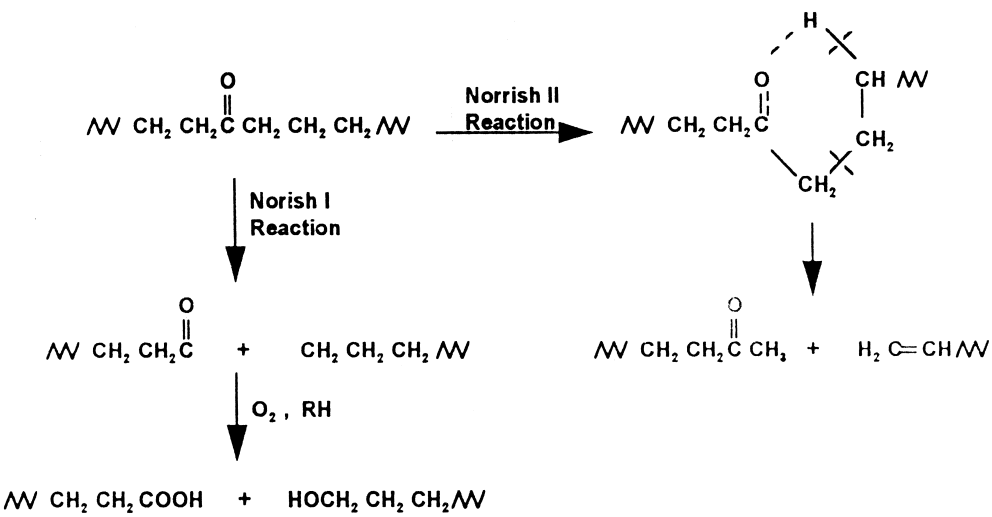


Fig. 9. Norrish I and Norrish II reaction mechanisms for the degradation of copolymers of ethylene.

methyl ketone end groups, and the latter can undergo further photochemical and oxidative reactions to form carboxylic acid end groups, which, again, should be susceptible to enzymatic interactions and oxidation if the fragments are of sufficiently low molecular weight.

Another method for photodegrading polyethylene is to include metal salts, which catalyze photooxidation reactions, in the solid polymer. The compounds most generally used for this purpose are divalent transition metal salts of higher aliphatic acids, such as stearic acid or dithiocarbonates or acetoacetic acid. The photochemical reaction is an oxidation–reduction reaction that forms free radicals capable of reacting with polyethylene, RH, to initiate an autooxidation chain reaction.

9. BLENDS OF BIODEGRADABLE AND NON-BIODEGRADABLE POLYMERS

The blending of biodegradable polymers, such as starch, with inert polymers, such as polyethylene, has received a considerable amount of attention for possible applications in the waste disposal of plastics. The reasoning behind this approach is that, in principal, if the biodegradable component is present in sufficient amounts and if it is removed by microorganisms in the waste disposal environment, the plastic or film containing the remaining inert component should lose its integrity, disintegrate and disappear. This concept has found its principal application in blends of minor amounts of starch with polyethylene in which the latter constitutes the continuous phase so that the blend can be melt processed to form films or plastics with polyethylene-like properties.

Granular starch, either in its virgin form or chemically modified on the granule surface to increase its compatibility with the matrix polymer, has been used to form these types of blends.[191–194] In a biologically active environment containing microorganisms that secrete amylases, the exposed starch granules on the surface of the sample and those granules within the sample which are in direct contact with the surface granules, can be enzymatically hydrolyzed and completely removed to create pits or voids. When a sufficient amount of the starch present in the blend is degraded and removed in that manner, the sample should lose its strength and or continuity and disintegrate. However, this effect occurs only for samples containing fairly large amounts of starch, of the order of 30% by volume, and polyethylene plastics and films containing so much granular starch have substantially decreased tensile, tear and impact strengths. That is, the effective connectivity and accessibility of the starch granules, which is required for extensive enzymatic hydrolysis and removal, is achieved only at relatively high starch contents. At lower starch contents, with blend compositions much below the threshold level for the connectivity of granules, very little effect on mechanical properties results from the biodegradation of the accessible starch component.[195,196]

The melt blending of nonbiodegradable thermoplastics, such as polystyrene, poly(vinyl chloride) and polyethylene, with biodegradable thermoplastics is receiving increasing attention. The biodegradable polymers that have melting temperatures which permit melt blending in a reasonable temperature range are primarily polycaprolactone[197] which has a melt transition at 60°C, and the bacterial copolyesters containing 3-hydroxybutyrate and 3-hydroxyvalerate units[198] which melt between 80 and 180°C depending on their repeating unit compositions. The rates of biodegradation of such blends can vary widely with variations in compatibility of the two components and in the blending procedure. These parameters

greatly effect the morphology of the resulting blend, which in turn controls the accessibility of the biodegradable component and, therefore, its biodegradability.[198–200]

9.1. *Polyethylene and starch blends*

Most synthetic polymers are considered to be resistant to microbial attack. Their biodegradability depends on various physical and chemical properties. Low molar mass polyethylene facilitates fungal growth to a certain degree.[201]

Griffin[202] reported on the degradation of buried LDPE in composting rubbish environments. Evidence was offered linking the initial stage of breakdown with the transfer by diffusion of unsaturated lipids from the compost to the polymer associated with the generation of peroxide by autooxidation.

Severini *et al.*[203] studied the environmental degradation of a stabilized LDPE film for up to 12 480 h of exposure. The highest concentrations of carbonyl and vinyl groups are observed between 8000 and 10 500 h of exposure. In the same time range the mechanical properties undergo the most significant reduction, while M_w and the thermal oxidation strength reach their lowest values. A spontaneous tearing of the film starts after about 10 000 h of exposure. The areas close to the tearing lines show the highest values of the relative optical densities of the carbonyl and vinyl groups. The crystallinity remains steady. The proposed degradation mechanism is based mainly on reactions of the Norrish I type. Severini *et al.*[204] studied the environmental degradation of a stabilized LDPE film to show that a reduction in mechanical properties and structural changes is observed after 8500 h of exposure.

Although several microorganisms facilitate the biodegradation of hydrocarbons, the biodegradation of polyethylene is somewhat slow.[205] Corbin and Henman[206] studied the biodegradation of ^{14}C-labelled polyethylene, where the rate of conversion was only about 2% per year. This suggests that the PE film could not be degraded significantly. This is because the degradation mechanism for linear hydrocarbons involves the oxidation of the terminal methyl group to a carboxylic acid group and the degradation of the resulting fatty acid by stepwise β-oxidation (two carbon units at a time). In high molar mass linear polyethylene, there are only two methyl groups which are not located in the bulk of the hydrophobic medium and these are not readily accessible to the microorganism. On the other hand, if ester groups could be introduced into the backbone of polyethylene, it would become more biodegradable.

Albertsson[207] studied the microbial and oxidative effects in the degradation of PE. Biodegradative conversion of ^{14}C present in the HDPE film to respiratory $^{14}CO_2$ during a two-year aerated cultivation with soil or with *Fusarium redolens* dropped from 0.36% by weight to less than 0.16% by weight when the HDPE film was deprived of most of its low molecular weight components by extraction with cyclohexane. A decrease of $^{14}CO_2$ production after extraction was observed in different abiotic aging cultures. This is direct evidence for the primary utilization of the short-chain oligomer fraction of the main crystalline material. The extractable oligomeric fraction of HDPE was analyzed by GPC, and M_n values of 1049, 1088 and 1297 were found in untreated, aged and biodegraded material, respectively, indicating that microbes can oxidize somewhat longer polyolefin chains than abiotic forces do during aging. The limited degradation of HDPE confined to the extractable materials is comparable to the degradation of straight-chain *n*-alkanes and presumably proceeds according to a similar mechanism. Such materials (*n*-alkanes) can exist in the interstitial spaces between the crystalline lamellae as fringed micelles which infiltrate these cavities as amorphous clusters. They

are also produced to some extent during aging and weathering. Protection of HDPE by antioxidants (sterically hindered phenols) resulted in an inhibition of microbial catabolism of ^{14}C to $^{14}CO_2$. Aging was also suppressed in this way, indicating that although remnants of the supported CrO_3 polymerization catalyst are responsible for a slight but cumulative abiotic oxidation of the unprotected polymer, this effect will be counteracted by the antioxidative additives. As biological degradation is superimposed on the chemistry of aging, a mutual synergism between the effects is feasible.

Colin *et al.*[208] studied the biodegradation of polyolefins and polyamide films under soil burial conditions extending for up to about three years. Based on the results of changes in elongation at break, the films have been ranked in the order of increasing sensitivity to degradation as: polyester = polypropylene $<$ LDPE = HDPE $<$ nylon-66. The degraded nylon-66 and PP films were characterized by IR, luminescence spectroscopy, and scanning electron microscopy, as well as by wet analysis for hydroperoxides. It was shown that the oxidative process was the cause of biodegradation for the nylon-66 film during soil burial.

Albertsson and Karlsson[209] studied the three stages in the degradation of polymers taking polyethylene as a model substance. Data relating to the degradative conversion of ^{14}C present in the LDPE film to respiratory $^{14}CO_2$ during a 10-year aerated cultivation with soil were presented. The degradation was performed with two sets of LDPE samples, one without additive (PE) and other containing UV sensitizer (NDPE). Samples were exposed to UV irradiation for 0, 7, 26 and 42 days. The degradation is characterized by three stages: (I) a constant degradation rate, (II) a parabolic decline in the rate of degradation, and (III) a subsequent final increase in the rate of degradation. The first step (I) is probably dependent on the environment. The material changes rapidly until some kind of equilibrium with the environment has been achieved. CO_2 is evolved, oxygen uptake is rapid, and a rapid change in mechanical properties is also observed. The second step (II) is characterized by low oxygen uptake, a low evolution of CO_2 and slow changes in mechanical properties, crystallinity and molecular weight. The change in mechanical properties are more or less lost due to the final collapse of the structure. For an inert material such as PE, 10 years is a relatively short time, so that only small indications for step (III) and a coming mineralization point can be observed. The changes are more evident for NDPE. The use of degradable materials, for example polypropiolactone, however, means that it is possible during a two-year period to study all three stages.

Karlsson *et al.*[210] studied the biodegradation of polyethylene and the influence of surfactants. LDPE samples were exposed to UV radiation for 27 days. Thereafter they were added to an abiotic and a biotic environment. By increasing the amount of polyethylene added to a soil system it was possible to compare how different amounts of irradiated PE affect the degradation rate and the evolution of CO_2. The degradation rate seems to be independent of the amount of PE added to the soil system but dependent on the biotic activity. The amount of CO_2 evolved follows close to a linear relationship. The addition of a surfactant to a nutrient solution containing PE results in an increased degradation rate. In contrast to the behaviour of similar samples without surfactant, this sample sinks to the bottom of the flask instead of floating on the surface of the solution.

Biodegradation of polyethylene was also characterized[211] using DSC and FTIR techniques. *Aspergillus niger*, a fungus, was used in the degradation of commercially available thermoplastic polyethylene samples. Quantitative calorimetric measurements performed on abiotic and biotic treated polyethylene samples revealed that the amorphocity of the samples

decreased during the biodegradation. In addition, it was found that the external substrates (sucrose) in the growth medium influenced the biodegradation process of polyethylene. Furthermore, the crystallinity data on different biotreated samples indicated that the adapted microorganisms were able to metabolize a small portion of polyethylene. The significance of the FTIR results for the polyethylene samples have been discussed.

Biodegradation of natural polymers such as cellulose and proteins during exposure to soil have been well understood.[212,213] In a review[214] concerning the environmental degradation of synthetic polymers under the influence of living organisms, it was found that the loss of strength, transparency, or good dielectric properties could be the result of biodegradation.

It was shown that lower molecular weight, normal (straight-chain) paraffins can be readily degraded by microorganisms, whereas their branched isomers showed slower degradation rates.[215–217] The ability of microbial enzymes to oxidize the terminal groups in hydrocarbon polymers has been studied by several researchers.[218–225] Potts *et al.*[225] compared the biodegradability of linear and branched hydrocarbons in the molecular weight range 170–620, using the ASTM test method D 1924-63. The linear hydrocarbons did not support the growth during the first three weeks of the test; also, none of the branched-chain hydrocarbons supported growth of the test fungi.

Polyethylene is one of the most resistant packaging materials. Intensive research is in progress to produce a decomposable grade. For biodegradation to take place it should be mixed with organic substances that help it to decompose more readily. To distinguish between the carbon dioxide formed by decomposition of the plastic and that formed from the readily decomposable substances, polyethylene containing the radioactive isotope ^{14}C has been used in a series of experiments in actual soil tests.[226]

Potts *et al.*[227] also studied the biodegradation of hydrocarbon samples and polyethylene after thermal degradation with four different strains of fungi and estimated the mycelium growth. Another fungus, *Aspergillus oryzae* was reported to grow on the surface of polyethylene and led to an increased degree of crystallinity.[228] The growth on polyethylene has been interpreted as being limited to the microbial action on the surface of an inert support without impact on the polymers.[229–232] However, it was found[233,234] that polyethylene is successively degradable in compost and the decrease in tensile strength is related to biodegradation. Biodegradation of ^{14}C-labelled HDPE has been studied[235] in the presence of cultivated garden soil (using several strains of white-rot fungi and the soil mould *Fusarium redolens*) by a specific respirometric technique.

Mobilization of ^{14}C from randomly labelled HDPE by the soil indicated a limited but not negligible degradation when the polymer films were exposed to strongly aerobic cultivation for more than two years.[236] These results did not resolve the controversial question of whether polyethylene is successively biodegradable or is completely withstanding any biological impact, or whether the chemical cleavage is enhancing the biological effect. In another study,[237] labelled HDPE in powdered form was used as a substrate exposed to attack by fungi, namely *Fusarium redolens*. This powder is the first polymeric solid product used in the industrial processing of polyethylene and is the only one of its kind reported to be biodegradable.[238] Many other studies to synthesize polymers that are more susceptible to biodegradation have been reported.[239–245]

According to Baum and Deanin,[246] photodegradation of polymers is essential before biodegradation. Some additives like transition metal complexes and the pure oleic ester of octanol[247] enhanced the photodegradation processes while others which are inherently

biodegradable increased the biodegradation of the polymer substrates.[9,248] In such cases, biodegradation was studied by observing microbial growth, or by measurements of physicochemical changes of the substrates in addition to oxygen uptake or of carbon dioxide production. These methods seem to suffer from the disadvantage that they do not distinguish between the degradation of the polymer and its additives. Instead, a scintillation assay method for carbon dioxide evolution has been used to study biodegradation.[249]

Several experiments have demonstrated the extent to which photodegradation of an additive can accelerate photochemical oxidation.[174] Photodegraded polyethylene films with and without the added photodegrading enhancers were exposed to microbiological attack in laboratory tests. The materials were inoculated with pure cultures of fungi, bacteria, and actinomycetes, previously isolated from polyethylene materials, in mineral media for up to a year. The effect of the microorganisms on the test materials was measured by determining the microbial growth and weight loss of the samples. The degree of microbial growth increased with the increasing degradation of polyethylene. Some weight loss occurred within the first seven weeks of the incubation period and this was due to the removal of additives. Furthermore, a considerable weight loss was caused by microorganisms only in samples which were irradiated by using a xenon lamp for up to 900–1200 h. Microorganisms were able to assimilate only low molar mass fractions of the polyethylene molecules comparatively quickly, thereby accelerating the chemical degradation.

Several plastics when physically or chemically degraded to brittleness, tend to be easily degraded by microorganisms, leaving CO_2, water, and other harmless substances at the end of the process.[9,250] This theory of complete microbial degradation of brittle, solid materials was not confirmed, however, by experiments. Photooxidative degradation of a quenched LLDPE sample, medium-density polyethylene (MDPE), and two kinds of HDPE films was studied using a medium-pressure mercury lamp.[251] Larger amounts of crosslinking and the build-up of oxidation products were found in LLDPE than in the other samples. The primary products of interaction between dienes and oxygen were involved in the initiation of the photooxidation reaction. Using FTIR difference spectra, the branch concentrations in the photoirradiated polyethylene samples was determined. Oxidation damage at the boundary region between the crystalline and amorphous phases was considered to be important in determining the embrittlement time.

Weiland *et al.*[252] studied the biodegradability of thermally oxidized PE under various conditions: (1) on solid agar in the presence of a suspension of mixed spores of four fungi (*Aspergillus niger*, *Penicillium funiculosm*, *Paecilomyces variotii* and *Gliocladium virens*); (2) in three composting units differing in temperature, moisture content and the nature of the composted materials; and (3) in liquid media (respirometric flasks) in the presence of three Streptomyces strains (*S. badius*, *S. setonii*, *S. viridosporus*) or of a suspension of microorganisms from compost. Qualitative evidence for the bioassimilation of the oxidized PE films was obtained with fungi and in composts. Coverage of the film surface by fungi increases as the molecular weight of the PE is decreased and reaches 60% when the initial M_n lies between 1500 and 600. With fungi and in compost, significant surface erosion was detected by SEM for samples with initial M_n values around 1000. Important changes were also observed by FTIR, DSC and GPC. This last method revealed in all cases the formation of a high molecular weight fraction that was not present before incubation with the microorganisms and a shift of the whole curve toward higher molecular weights. This is evidence of chain condensation probably due to thermal reactions at the low partial pressure of O_2 prevailing in the industrial

composting units used in this work. It is probably accompanied by bioassimilation of the lower molecular weight fractions. For incubations performed in liquid media in the presence of a suspension of microorganisms from compost, biodegradation was significant when the substrate concentration was very low (0.006%). Despite the presence of unavoidably large errors in these conditions, oxygen uptake was evident and the biodegradation of the lower molecular weight fraction of the sample was clearly demonstrated.

Guiller *et al.*[253] have studied the biodegradability of photodegraded plastic composites wherein the resistance of conventional plastics to microorganisms is attributed to two factors: surface areas and a relative impermeability of plastic films and moulded objects, and the very high molar mass of the plastic materials. Microorganisms tend to attack the ends of large molecules, the number of ends being inversely proportional to the molecular weight. In order to make plastics degradable, it is thus necessary to first break them down into very small particles with a large surface area and, secondly, to reduce their molecular weights.

Degradable plastics are materials designed to be broken into smaller components or to disintegrate and eventually be converted to nonharmful substances over predetermined periods of time and under average environmental conditions. Polyethylene is resistant to attack by microorganisms or by chemical means other than photooxidation in most naturally occurring environments.[214,254] In order to increase its degradation, a number of approaches are being used such as copolymerization with ketone-containing materials, compounding with metal salts, starch, and other additives.[255,256]

Long-term studies on the biodegradation of ^{14}C-polyethylene films show less than 0.2% (by weight) evolution of CO_2.[158,207,236,257] The effect of additives such as UV-sensitizers[209] and *N*-dotriacontane[249] have shown a five-fold increase and an initial increase followed by a slow down, respectively, in the conversion to CO_2.

Cellulose, certain proteins, and rubber are natural polymers long exploited industrially as load-bearing solids, and are all capable of biodegradation either by direct enzyme scission or, in the case of natural rubber, by oxidative scission followed by biological metabolization.[258] Starch, a natural polymer of comparable industrial standing as judged by tonnage production, has not been regarded traditionally as an element of the plastics industry despite its comparable mechanical properties, attractive economics, and useful particulate form. The urgency of providing materials with low energy demands from renewable resources, which also offer biodegradability as an option in waste disposal not conflicting with recycling or incineration activities, has served to promote starch blends and alloys in polymer applications.

The structural and mechanical properties of extruded high-amylose and normal cornstarch were studied as a function of time and humidity to determine the suitability of high-amylose cornstarch for use in biodegradable plastic materials.[259] After extrusion at 170°C and 20–30% moisture, high-amylose starch was mostly amorphous, with small amounts of V- and A-type crystal structures. Tensile strengths for the extruded high-amylose starch ribbons were rather stable with time (65, 50, and 35 MPa at 20, 50, and 80% RH) and were higher than those for normal cornstarch (25, 40, and 15 MPa after 84 days at 20, 50, and 80% RH). Elongations at break declined gradually with time for the high-amylose starch (6, 11, and 11% after 84 days at 20, 50, and 80% RH), while rapid declines were seen for normal cornstarch at higher humidities (3, 9, and 3% after 84 days at 20, 50, and 80% RH). Differential scanning calorimetry revealed that normal cornstarch aged at a high humidity had much larger sub-T_g endotherms than high-amylose cornstarch. These endotherms reflect decreases in enthalpy and free volume which occur in amorphous polymers due to structural relaxation.

It appears, therefore, that plastic materials prepared from gelatinized or melted high-amylose cornstarch should have greater strength and flexibility and slower physical aging than those prepared from gelatinized normal cornstarch.

The ASTM D5210-91 protocol for evaluating the biodegradability of a polymer was examined.[260] The reactor design was modified not only to account for the total CO_2 evolved but also to allow for the simultaneous carbon assessment in microbes, soluble products, and solid samples. Improvements in the test procedure were implemented such as (1) refining the CO_2 pretrap and posttrap design, (2) optimizing the carbon dioxide removal efficiency, (3) accounting for the total polymeric carbon, (4) standardizing the inoculum, and (5) revising the nutrient medium. By growing the sludge on a suitable substrate prior to polymeric exposure, a constant microbial density was obtained. The modified ASTM method provides an assessment of the polymeric carbon degradation at any given time. The results of this work have specific significance to the behaviour of polymers in a sewage waste treatment plant, where sludge is continuously being aerated, and also for aerobic biodegradation in general.

Incorporation of starch into a polyolefin matrix was first proposed[261,262] as an effective means of accelerating the deterioration of plastics under biotic environmental exposure conditions. The inclusion of starch, a readily biodegradable biopolymer, into the synthetic polymer is believed to result in rapid enzymatic hydrolysis of the starch under biotic exposure conditions, leading to a void-containing matrix. the reduced mechanical integrity of the ensuing void-containing matrix leads to its facile deterioration, and perhaps even promotes subsequent biodegradation of the synthetic polymer, due to increased surface area available for interaction with the microorganism.

Physical incorporation of granular starch derivatives as functional additives and fillers into polyolefins during polymer processing, e.g. foaming, extrusion, injection moulding or film blowing on existing equipment is well known and has been used for many years. In the absence of additives, films made from starch or amylose are brittle and sensitive to water (hygroscopic).

A water dispersion of corn starch and a plasticizer, such as glycerol, can be cast into a clear flexible film. However, these films have no industrial potential, as nonsupported films, because they deteriorate in water and become very brittle under ambient conditions. The formation of blends of two or more polymers is an obvious technique to tailor materials for specific applications. McCarthy *et al.*[263] studied the processing and blend morphology effects on biodegradability. This study focuses on blends of biodegradable polymers and the effect of various blending and processing techniques on the biodegradation rates as measured upon exposure to two disposal environments, aerobic composting and soil burial. Otey *et al.*[10] found that film quality is greatly improved by adding PVA to the starch–glycerol formulation, especially when a water-resistant coating was applied. Since early 1975, one company has been using combinations of modified starch and PVA to produce water-soluble bags.

Otey *et al.*[2] studied biodegradable films from starch and ethylene–acrylic acid (EAA) copolymers with the aim of improving their water resistance and preventing them from becoming brittle with age. Compatible mixtures containing up to 90% starch and EAA copolymer were milled or cast from aqueous dispersions into flexible, nonsupported films without the aid of a conventional plasticizer. These films were water resistant and appeared to have acceptable physical properties for a variety of packaging and agricultural mulch applications. Cast films, with up to 50% starch and 2% paraformaldehyde, resisted outdoor weathering for more than 2 months. The reasons that these films remain flexible with aging

is not clearly understood. However, it seems possible that starch molecules are expanded and quite flexible when first cast from an aqueous dispersion, but upon drying they contract and various bonding forces cause them become brittle.

The added EAA may be associating with the starch molecules enough to hold them in their expanded, flexible states. As the amount of EAA is decreased, this association can be partially disrupted, especially with age or in the presence of a solvent such as water.

A technique for blending gelatinized starch and poly(ethylene-*co*-acrylic acid) (EAA) to produce flexible blown films that contain high levels of starch was introduced by Otey *et al.*[12] Ammonia and 2–10% moisture were essential ingredients for obtaining uniform films. The inclusion of polyethylene in the film formulation improved the economics and increased the UV stability and the rate of biodegradability of the blown films. The films have potential applications as agricultural mulches and packaging, especially where biodegradation is important. Films made from this formulation had no observed permeability characteristics. Otey and Westhoff[264] found the films to be much more transparent and have promising semipermeable characteristics when strong alkalis such as sodium hydroxide were used in place of ammonia. Observed permeabilities for six solutes were determined by the use of a rotating dialysis cell. After 1 h exposure of 1.5% solute concentration (w/w), urea diffused through one film 7.6 times faster than sucrose. Increasing starch levels or incorporating water-soluble compounds into the films significantly increased diffusion rates.

Urea and polyols were added to starch–EAA formulations to facilitate the preparation and to improve the economics and quality of the starch-based films.[190] The principal benefit of the urea was to improve the gelatinization of starch at low levels of water, thus allowing direct extrusion of a uniform film from a semidry blend (~16% H_2O) and avoiding the need to premix starch–EAA in a heavy-duty mixer with large amounts of water prior to extrusion processing. Initial tensile strengths of the urea-containing film were generally lower than those made by the premix method, but after water soaking to remove the urea, the tensile strengths were nearly equal to those made without urea. Glycerol and starch-derived polyols can be added to starch–EAA systems to increase the percentage of the biodegradable component without adversely affecting the physical properties of the films.

The biodegradation of PE films containing (by weight) 40% gelatinized cornstarch and 15% EAA was studied in a variety of aqueous environments.[265] In the laboratory, some amylolytic bacteria degraded starch in the film more rapidly and to a greater extent than others during a 60-day incubation. Loss of starch was accompanied by a concomitant decrease in film tensile strength, facilitating the disintegration of the film from mechanical stress. Films composed of PE–EAA and PE–EAA–starch were exposed to three fresh water ecosystems (river, marsh, pond) for 90 days. The surfaces of all three films were rapidly coated by a complex biofilm containing bacteria, algae, fungi, protozoa and diatoms. These data suggest that microbial starch degradation, mechanical disintegration, and biodisintegration are all factors that influence the environmental fate of starch-containing plastics.

PE films containing (by weight) 40% gelatinized cornstarch and 15% EAA were found to exhibit a heterogeneous microstructure, with starch distributed nonuniformly throughout the matrix.[266] Comparatively little starch was exposed on the film surfaces. Internally, layers of starch-rich and starch-deficient regions were evident. Surface accessible starch was rapidly degraded by amylopectin bacteria, while internal starch was degraded much more slowly. Some internal starch was highly resistant to amylopectin degradation. In model studies, amylose formed a tight complex with EAA that was not degraded by amylases. These data

suggest that the existence of hydrophobic barriers by amylose–EAA complexes in high-starch-containing plastic films influences the biodegradation properties of these films, and that these features may be exploited for regulating biodegradability as a value-added property.

X-ray diffraction, ^{13}C NMR, DSC, FTIR and fluorescence microscopy have all been used to study the structure, compatibility, and morphology of films made from starch, EAA, and PE before and after exposure to a mixture of highly amylolytic bacteria.[267] The components of starch, amylose and amylopectin interact with EAA via the formation of V-type inclusion complexes and hydrogen bonds. PE appears to be immiscible with the starch–EAA complex, with each forming sheet like domains. The amylopectin in the films is susceptible to digestion by the bacterial consortium while the crystalline EAA–amylose complex is resistant. Digestion begins at the film surface and then proceeds inwards with sheet like areas of starch removed. The good compatibility between starch and EAA as well as the migration of EAA to the film surface explains the resistance of such films to digestion by conventional amylases.

The effects of the marine environment on unstabilized PE–starch composites, with or without a metal catalyst (MC) and autooxidant (AO), has been studied.[268] Starch tends to absorb water. For PE–starch composites containing MC and AO exposed under plain seawater, there appears to be practically no microbial activity as indicated by no surface erosion and no change in tensile properties. However, the decrease in molecular weight on the surface indicates ongoing chemical degradation due to the presence of MC and AO. For PP–starch composites with no additives exposed to plain seawater, there seems to be no microbial activity or chemical degradation. However, for PP–starch composites containing MC, AO or plasticizer (PL) exposed under soft mud, surface erosion due to microbial activity is evident.

An ideal film would degrade at the end of the crop season to such an extent that it would not foul tillage equipment and would completely degrade in a short time when buried in the soil. The rate of biodegradation of plastics may be increased by including a highly biodegradable polymer in the plastic matrix. Starch, being readily biodegradable, could function as the labile component when incorporated into PE-based mulch films. The authors discussed the formulation of the films. It is concluded that the potential of these films as agriculture mulches depends on their total performance, which includes cost as well as efficacy. A practical formulation of 40% starch, 20% EAA, 25% LDPE and 15% urea would have a material cost of 35–40 ¢/lb, which is near the price of LDPE.[269] If 230 lb/acre of LDPE were applied, the cost of removal (100/acre) would be 43 ¢/lb of film. Hence considerable economic benefit could be realized by mulching crops with degradable films that need not be removed from the field.

The use of starch additives in PE and EAA has been applied commercially in the manufacture of bags. Compatibility problems are minimized by use of silane coupling agents. In theory the polymers in contact with soil or water are attacked by microbes which ingest the starch additives in the polymer matrix. This leaves behind a porous sponge like structure with a high interfacial area and a low structure integrity. Thus the starch component is ingested first, followed by enzymatic attack on the polymer. This attack consists of many consecutive enzymatic reactions. In each cycle of attack, one base unit, usually an acetic acid molecule, is split off. Thus the average molecular weight tends to decrease relatively slowly.[85]

Starch has been used for many years as an additive to plastic formulations for various purposes. Griffin[192] reported starch as a biodegradable filler in LDPE. The interesting observation that degradation of the polyolefin component of the starch/LDPE composites

was observable in composting municipal solid waste (MSW) but not during simple garden soil burial led to the realization that some component of the MSW must be responsible.[202] Chemical examination of the LDPE films retrieved from composted waste at municipal facilities revealed that the active agent was an unsaturated cooking oil which was selectively absorbed by the LDPE under the warm conditions of the Dano process. Up to 9% of rancid fat rich in peroxides could be found in these films and this could explain the degradative activity in terms of oxidative chain breaking followed by biological attack on the resulting fragments. The criteria for achieving biodegradation of polyethylene was set out very clearly by Hueck[270] and it appeared that the presence of readily oxidisable unsaturated materials in the polymer formulation could achieve these criteria. Furthermore the possibilities of enhancing biodegradability by making mixtures of degradable and resistant substances were mentioned by Gorzynska.[271] Accordingly, the final formulation of the original degradable films marketed by the Coloroll company emerged as a composition including about 7% of hydrophobic maize starch plus a small percentage of the pure oleic ester of octanol. Under moist conditions, contact with earth would introduce into the discarded films sufficient transition metal contamination to trigger the autooxidation of the unsaturated fatty additive. Since then many studies have been performed on the degradation of PE–starch composites.

Albertsson *et al.*[272] studied the spectroscopic and mechanical changes in irradiated starch-filled LDPE. LDPE containing corn starch (3.9%, 5.8% and 7.9% by weight) without further additives and corn starch in a prooxidant formulation (= masterbatch (MB): 10, 15 and 20% by weight), were analyzed by FTIR and draw testing after irradiation for periods of up to 500 h. The carbonyl index for LDPE–20% MB after 500 h irradiation was 2.5 times the value obtained in pure LDPE and LDPE–starch. The hydroperoxide index for the same material was three times the value obtained in pure LDPE and in LDPE–starch.

During irradiation, the tensile strength decreased to almost the same extent for all three materials—a mean value of 70% of the initial value was obtained after irradiation. The elongation at break for pure LDPE decreased during irradiation from 650 to 500%. After irradiation the decrease was considerable—a mean value of 60% being recorded for the elongation at break of LDPE–MB.

A material susceptible to photolysis is thus obtained by the addition of corn starch and a master batch containing LDPE, styrene–butadiene copolymer (SBS) and manganese stearate. Incorporating only starch into LDPE did not significantly change the susceptibility of the material to photolysis. LDPE–MB is thought to degrade by an initial photooxidation (auto-oxidation) process and a subsequent attack by microorganisms (i.e. biodegradation). Further ongoing studies on the biodegradation of pretreated LDPE–MB will be reported elsewhere. The induction time is accelerated by pretreatment. Irradiation or thermal treatment generates free radicals causing autooxidation which should facilitate a later biodegradation.

Goheen and Wool[196] studied the degradation of PE–starch blends in soil. Binary polymer films containing different percentages of corn starch and LDPE were exposed to three different types of soils over a period of eight months and monitored for starch removal and chemical changes of the matrix using FTIR spectroscopy. A standard curve using the area of the C–O stretch band and an empirical second-degree polynomial to fit the data made it possible to calculate the starch concentration over a wide range (0–46% by mass). Starch removal was found to proceed rapidly during the first 40 days and to near completion in very high starch blends (52% and 67% by weight). Starch removal was slower, consisting of mostly surface removal in 29% starch blends. Weight loss data showed similar gross features.

Weight loss and spectroscopic data were consistent with percolation theory and suggested that starch removal continues past 240 days. Degradation rates in different soils containing different amounts of organic matter were approximately the same after a period of a few weeks. IR analysis did not show significant chemical changes in the polyethylene matrix after 240 days. However, the matrix did show evidence of swelling, an increase in the surface area and the removal of low molecular weight components when submerged in seawater.

Breslin and Li[273] studied the weathering of starch–polyethylene composite films in marine environments. Polyethylene and starch–PE composite films were exposed outdoors in the strawline of a marsh and in seawater on flow-through sea tables in the laboratory. The deterioration of these films following exposure was measured by determining the changes in tensile properties, weight loss, starch loss, and carbonyl content of the sheet plastic films. Low rates of deterioration were observed for the control and starch–polyethylene composite films submerged in seawater. In contrast, both the starch–PE composite and control PE films rapidly deteriorated during exposure in the strawline of a marsh. Differences in the observed rates of deterioration of the films placed in the exposure sites were attributed primarily to photodegradation of the films placed in the strawline of a marsh.

Peanasky *et al.*[274] investigated the accessibility of starch in PE–starch blends by computer simulation, percolation theory, and acid hydrolysis experiments. The object of this work was to model the bilateral invasion of microbes in PE–starch blends as a function of starch concentration (p), and thickness of the material. It was found that computer simulations in three dimensions were in agreement with both percolation theory and the acid digestion experiments. In computer simulation, the accessibility is highly dependent on the percolation threshold concentration (p_c), which is 31.17%. Similarly, the accessibility of starch is highly dependent on an apparent percolation threshold near 30% by volume or approximately 40% by weight of starch. At $p < p_c$, a small amount of starch is removed from the surfaces only, but at $p > p_c$ connected pathways existing throughout the bulk of the material facilitate large amounts of starch extraction. The sharpness of the transition at p_c increases with the ratio of sample thickness to starch particle size. The results of this work have application to conduction and reacting systems where one component is dispersed in a matrix of the other.

Willett[275] studied the mechanical properties of LDPE/granular starch composites as functions of starch volume fraction, granular size, and presence of compatibilizer. Property–volume fraction relationships were interpreted using various theories of composite properties. The dependence of elongation ($\epsilon \sim \phi^{1/3}$) and tensile strength ($\sigma \sim \phi^{2/3}$) agree with theoretical predictions, although the proportionality constants are less negative than theoretical values. The addition of compatibilizer (EAA) did not significantly affect the elongation or tensile strength, but significantly increased the composite tensile modulus. The cornstarch/PE moduli could be described by the Kerner or Halpin–Tsai equations. Analysis of the composite moduli data using the Haplin–Tsai equation allowed the estimation of the modulus of granular starch. The value obtained, 15 GPa, is considerably greater than most unfilled synthetic polymers of commercial importance, but significantly lower than the modulus of cellulose. It is also greater than a previously reported value of 2.7 GPa.

Tsao *et al.*[276] studied the influence of soil macroinvertebrates on primary biodegradation of starch-containing polyethylene films. The primary biodegradability of PE films containing different percentages of cornstarch (0–50%) and other additives (prooxidant, oxidized polyethylene) was tested using four species of earthworms (*Eisenia fetida*, *Lumbricus terrestris*, *Aporectodea trapezoide* and *Aporectodea tuberculata*), three species of cockroaches

(*Periplaneta americana*, *Bleberus* sp., *Blattella germanica*), termites (*Reticulotermus flavipes*), sowbugs (*Porecllio laevis*), and crickets (*Acheta domesticus*). These studies were conducted to elucidate the potential role of soil macroinvertebrates in degrading starch/PE biodegradable plastics. The results of the macroinvertebrate bioassays indicate that crickets, cockroaches, and sowbugs consumed starch-containing PE films most readily. In addition, the degree to which the films were attacked and consumed was directly related to the starch content of the film. Films with oxidized polyethylene and those containing prooxidants (vegetable oil and transition metal catalysts) were also consumed. None of the four species of earthworms tested or the termites showed any activity toward the starch/PE films. These results have important implications for determining the fate of novel plastic formulations which claim to be biodegradable in natural environments. Studies such as these, coupled with studies on microbial degradation, will help provide the type of information needed to assess the environmental fate of biodegradable starch/PE plastics and will help fill voids in the scientific database regarding this rapidly developing field.

Urea in the form of pellets is the major synthetic fertilizer used in rice fields. However, only half the urea applied in this manner is estimated to be absorbed by the rice plants; the other half is lost, causing water contamination. Various technologies are currently being explored to control urea release in order to maximize absorption and thereby, simultaneously, reduce contamination of the environment. One such technology is encapsulation of the urea in a permeable film that is designed to release the urea at a rate conducive to plant growth.[277] One important consideration in the development of this technology is the cost of the permeable film. LDPE is one of the least expensive polymers, and it was therefore selected as the first candidate for urea encapsulation. Another advantage of using LDPE is its ability to photodegrade in sunlight. Degradation is known to be enhanced by the addition of starch particles. Starch degradation via exposure to soil bacteria would be expected to develop micropores in PE/starch films. The microporous film would then be capable of enhanced urea release.

To test the suitability of LDPE films containing starch for modified temperature packaging of fresh food-stuffs, their permeabilities to O_2, CO_2 and N_2 gases were determined.[278] Films with 10, 15 and 20% mass of starch were compared against plain LDPE films at different temperatures. The observed CO_2/O_2 permeability ratio suggested an advantage of the starch-modified films over plain LDPE films. The results also indicated a temperature dependence of permeability based on the Arrhenius equation.

Albertsson *et al.*[279] studied the degradation product pattern and morphology changes as a means to differentiate between physical/chemical (abiotic) and biological (biotic) aged degradable polyethylene. Comparisons were made between LDPE, LDPE + 7.7% starch and LDPE + 20% (starch + prooxidant). Prooxidized samples were subjected to aqueous sterile and aqueous biotic (*Arthrobacter paraffineus*) environments at ambient temperatures for 15 months, and thermooxidation at 95°C in water. Carboxylic acids were identified in the abiotically degraded samples in contrast to the biotic environment, where assimilation of lower molecular weight products, especially carboxylic acids, had taken place as determined by gas chromatography and gas chromatography–mass spectrometry. Several hydrocarbons (C–C) were also present in these samples. This is in agreement with the proposed biodegradation mechanism of LDPE. The morphology changes, as monitored by X-ray diffraction (XRD) and SEM, were different in the two environments. A decrease in lamellar thickness (l) was demonstrated for biotically degraded LDPE + 20% (starch + prooxidant), while the

corresponding abiotically aged samples showed a constant or increased value of *l*. The crystallinity (XRD-w_c) for samples aged at ambient temperature showed that prolonged exposure to *A. paraffineus* resulted in decreasing value of w_c. In the accelerated environment, however, a constant increase in XRD-w_c was monitored. The principal difference between abiotic and biotic degradation of polymers is that microorganisms use polymers to gain energy. This is manifested as different degradation product patterns (reflecting degradation mechanisms) and a decreasing value of crystallinity and lamellar thickness with time. The abiotic degradation breaks bonds and releases degradation products, leaving the remaining polymer rearranged with a higher degree of order.

Shah *et al.*[280] characterized the initial degradation mechanism of starch-filled LDPE. Swelling of the starch in the starch–LDPE strips was observed when native starch was used. So, LDPE was compounded with well-dried, modified, granular starch (CATO-32) according to the Griffin technique. The starch and additive system was mixed with LDPE on a two roll mill at 125–130°C. A single screw Brabender extruder was used to obtain starch-filled LDPE strips. Accelerated degradation of the starch-filled LDPE strips was carried out using various laboratory tests (starch hydrolysis by α-amylase at 95°C, thermal oxidation in an air oven at 80°C and exposure to 254 nm UV radiation). Changes in the various properties of the strips during the course of degradation were evaluated using the following: universal testing machine (UTM) for mechanical properties, extrusion plastometer for melt flow index, SEM for surface morphology and IR spectrometry. SEM micrographs after starch hydrolysis show that α-amylase acts on the surface of the starch to cause cracks, holes, pitting and erosion which increase the surface area. The starch filled LDPE becomes brittle when it undergoes thermal oxidation. The prooxidant system (oleate and Fe) enhances the rate of thermal oxidation of the samples by 15–20%. An increase in the carbonyl and vinyl concentrations shows that chain scission reactions in the polymer chain were initiated by UV radiation. Thus various environmental factors have synergistic effects on the degradation of starch–LDPE.

A new method for evaluating the biodegradability of starch-based and certain other polymer blends uses the pre- and postexposure stable carbon isotope composition of material coupled with weight loss data to determine which components have degraded.[281] The naturally occurring stable isotope of carbon, ^{13}C, is enriched in corn starch (delta ^{13}C approximately −11‰) compared to petroleum-derived synthetic polymers (delta ^{13}C, approximately −32‰). Results for starch–synthetic polymer blends indicate that the delta ^{13}C signatures of these blends are near-linear mixtures of their component delta ^{13}C. Values of delta ^{13}C for starch–synthetic polymer blends exposed to biologically active laboratory soils and artificial seawater conditions are depleted in ^{13}C compared to unexposed samples, suggesting a loss of the starch component. Combined with weight loss data for the exposed samples, the delta ^{13}C values are statistically consistent with models requiring the loss of the soluble component glycerin, followed by loss of starch, then the petrochemical polymer, or the simultaneous loss of starch and the petrochemical polymer. Replicate delta ^{13}C analyses of starch–synthetic polymer blends increase the statistical power of this relatively inexpensive, accessible technique to discriminate between degrading components.

9.2. *Modified polyethylene and starch blends*

Graft copolymerization of thermoplastic polymers onto starch provides another method for preparing starch–polymer composites. An important advantage of graft copolymerization is

the fact that starch and synthetic polymers are held together by chemical bonding rather than existing merely as physical mixtures. The two dissimilar polymers therefore tend to be more intimately associated, and separation of the two polymer phases is less likely to occur. Fanta and Doane[282] have made an extensive study of the synthesis and properties of starch graft copolymers: in the course of this research, the properties of starch-g-poly(methyl acrylate) (S-g-PMA) have proven to be especially interesting. Graft polymerization of methyl acrylate onto either granular or gelatinized starch takes place readily in water with ceric ammonium nitrate initiation, and graft copolymers containing about 50–60% PMA can be easily prepared with minimal formation of ungrafted homopolymer. The combined properties of the rigid starch matrix and PMA ($T_g = 8°C$) result in the formation of a tough leathery plastic on extrusion processing. Dennenberg *et al.*[285] showed that the starch portion of these S-g-PMA extrudates is susceptible to fungal attack.

Patil and Fanta[283] prepared S-g-PMA copolymers containing 55–60% PMA from cornstarch, high amylose cornstarch, and waxy cornstarch with ceric ammonium nitrate initiation. The graft copolymers were characterized with respect to the percentage conversion of monomer to polymer, grafted PMA content, grafting frequency, molecular weight and molecular weight distributions of the PMA grafts. Variables investigated in the graft copolymerization reaction were nitric acid concentrations, ceric ion-to-starch ratios, reaction times, gelatinization of starch and reactant concentrations in water. At high concentrations, high conversions of methyl acrylate to grafted PMA could be obtained in less than 0.5 h at 25°C.

Henderson *et al.*[284] grafted PMA onto wheat starch by γ-irradiation and chemical initiation, respectively. The effect of water on the S-g-PMA extrudate, molecular weight distributions of the homopolymer, tensile and dynamic mechanical properties of the extrudate and moulded samples of both graft polymers were all reported.

Dennenberg *et al.*[285] prepared S-g-PMA copolymers having grafted side chains with molecular weights of less than 500 000. This material can be easily extruded into a film which shows excellent initial tensile strengths and elongations. Tensile strength, however, falls off rapidly after 70 h of water immersion at 25°C. S-g-PMA films show excellent susceptibility to fungal growth, some samples losing more than 40% of their weight after 22 days of incubation with *Aspergillus niger*. Tensile tests and SEM of the incubated samples, after being freed of mycelium, indicate substantial biodegradation of the starch portion of the copolymer. This material may have an application as a biodegradable plastic mulch.

Although Dennenberg *et al.*[285] confirmed that the starch portion of these graft copolymers is indeed susceptible to fungal attack, PMA is resistant to biodegradation. Enzymes produced by microorganisms can theoretically hydrolyze ester linkages to yield poly(acrylic acid); however, the biodegradation of poly(acrylic acid) is molecular weight dependent,[286] and high molecular weight polymer apparently remains resistant to microbial attack despite its water solubility.

To enhance the biodegradability of the PMA portion of the graft copolymers, Fanta *et al.*[287] introduced poly(vinyl acetate) segments into the polymer grafts by copolymerizing vinyl acetate with methyl acrylate during the grafting reaction. Esterases produced by microorganisms will convert poly(vinyl acetate) to poly(vinyl alcohol). Poly(vinyl alcohol) can then undergo further microbial attack to cleave the polymer chain,[288,289] thus yielding fragments to further degradation in the environment because of their low molecular weights. Bailey *et al.*[290] and Feterpeker *et al.*[291] enhanced the biodegradation of synthetic polymers by a similar approach in which degradable polyester segments were introduced into polymer chains via ring-opening polymerization of cyclic keten acetal comonomers.

An alternative approach is to bring about some compatibility of the starch and synthetic polymer by blending starch with polymers containing polar functional groups that can interact with starch. In recent years, several patents have been granted[292–294] and the Novon divison of the pharmaceutical giant Willett[294] used starch, copolymers of an olefin, and optionally, a poly(mono)olefin or poly(mixed)olefin to make blends that were injection moulded or film blown into commercial articles. An increase in the starch percentage adversely affected the physical properties of the blends.

Yet another economical and commercially viable approach is to form graft or block copolymers *in situ* during the blend preparation by using polymers containing reactive functional groups. The blending is performed under the conditions that promote the reaction. This method is commonly known as 'reactive blending'. Small amounts of blocks of graft copolymers formed during the blending process, due to reaction between the two components, are generally enough to stabilize the morphology and improve the properties of the blend. Reactive blending is known to improve the compatibility and interfacial adhesion of two immiscible polymers. This technique has been extremely popular in generating polymer blends in the synthetic polymer industry.[295] Synthetic polymers having functional groups such as carboxylic acid, anhydride, epoxy urethane, or oxazoline, can react with hydroxyl or carboxyl groups (in modified starch) to form a blend with stable morphology.

Jane *et al*.[293] in their patent used starch, oxidized polyethylene, and LDPE to produce films. According to these authors, the carboxy and ketone groups of oxidized polyethylene react with the hydroxyl groups on the starch to form bonds. They also report that as the percentage of starch in the blend is increased, the tensile strength and the percentage elongation decrease.

The reactivity of the functional groups is an important parameter in reactive blending. Most of the blends are commercially prepared in an extruder. The functional groups should react to form the required concentrations of graft or block copolymers in the short residence times typical of extrusion processes. From this point of view a cyclic anhydride group may react more quickly than the carboxylic group because of its higher reactivity. Unlike carboxylic groups, reaction of anhydride with hydroxyl to form an ester is not an equilibrium reaction as no water is produced during the reaction. Anhydride functionality can be incorporated into a polymer by copolymerization or grafting of anhydrides like maleic anhydride. Maleic anhydride can be grafted with relative ease onto many polymers under normal melt processing temperatures.[295]

Although studies involving reactive blending of starch with anhydride functional polymer are unavailable, there are indications that the anhydride compounds improve the properties of composites made from cellulosic fillers. Pholman[296] reported on the improved bonding between cellulose fibres using anhydrides. Maldas and Kokta[297] used anhydride functionalized polystyrene to improve the compatibility and adhesion with cellulosic fibres (wood chips and pulp). They reported that the properties of the composites varied with the concentration of maleic anhydride, type of wood used, and pulping technique. Maleated high density polyethylene (HDPE) improved the tensile strength of composites containing wood flour with increasing concentration of filler.[298]

Vaidya and Bhattacharya[299] studied the properties of blends of starch and synthetic polymers containing anhydride groups. Corn starch was blended with styrene maleic anhydride copolymer (EPMA), and the corresponding nonfunctional polystyrene and ethylene propylene copolymers. The concentration of starch in the blend was varied between 50 and 80% by weight. The torque generated during blending is reported as a function of starch content,

mixer speed, and mixing time. Torque increased with increasing starch content for starch/SMA blends; the reverse was true for starch/EPMA blends. The torque was higher for the blends of the anhydride functional polymers compared to blends of the corresponding nonfunctional polymers. Water absorption of the blends increased with an increase in the starch content. Starch/SMA blends made at higher mixer speeds or times were more water sensitive. Blends containing EPMA absorbed less water than SMA blends containing the same weight fraction of starch. The tensile strengths of blends containing functional groups were superior compared to the blends made from nonfunctional polymers. When the starch contents increased from 60 to 70%, the tensile strength remained unchanged for the SMA blend but increased for the EPMA blend. All samples supported the growth of microorganisms, which increased with increasing starch content.

10. APPLICATIONS

The applications of biodegradable polymers have been focused on three major areas: medical, agricultural, and consumer goods packaging. Some of these have resulted in commercial products. Because of their specialized nature and greater unit value, medical device applications have developed faster than the other two.

10.1. *Medical applications*

Biodegradable plastics have been developed as surgical implants in vascular and orthopedic surgery as implantable matrices for the controlled long-term release of drugs inside the body, as absorbable surgical sutures, and for use in the eye. Recently the term biomaterial was defined as a nonviable material used in medical device applications that is intended to interact with a biological system.[300] It is also important to define biocompatibility, which deals with how the tissue reacts to foreign materials. Biocompatibility is the ability of a material to perform with an appropriate host response in a specific application.[301] Biomaterials in general are used for the following purposes:

(a) to replace tissues that are diseased or otherwise nonfunctional, as in-joint replacements, artificial heart valves and arteries, tooth reconstruction and intraocular lenses;
(b) to assist in the repair of tissue, including the obvious sutures but also bone fracture plates, ligament and tendon repair devices;
(c) to replace all or part of the function of the major organs, such as in haemodialysis (replacing the function of the kidney), oxygenation (lungs), left ventricular or whole heart assistance (heart), perfusion (liver), and insulin delivery (pancreas);
(d) to deliver drugs to the body, either to targeted sites (e.g. directly to a tumour) or sustained delivery rates (insulin, pilocarpin, contraceptives).

10.1.1. *Surgical sutures*

Tissue damage which results in a loss of structural integrity, for example a deep cut in soft tissue or a fracture of a bone, may or may not be capable of unassisted self healing. The insertion of some material or device to hold the tissue together may facilitate the healing process. The classic examples are the use of sutures to hold both deep and superficial wounds together.

Once the healing is complete, the suture becomes redundant and can impose undesirable constraints on the healing tissues. It is preferable to remove the material from the site, either physically or by degradation.

Synthetic absorbable sutures were developed in 1960s, and because of their good biocompatibility in tissues, they are now widely used in tracheobronchial surgery, as well as general surgery. They are multifilament-type sutures, which have good handleability. Polyglycolide (PGA), poly-L-lactide (PLA) and their copolymers, and polyglactin are the most popular and are now commercially available. However, for continuous suturing, braided sutures with nonsmooth surfaces are not useful. Monofilament sutures have smooth surfaces and are adequate for continuous suturing. For a monofilament suture, PGA or PLA are too stiff and inflexible. The more flexible polydioxanones and polyglyconates can be used as sutures because of their lower bending moduli. Furthermore, copolymers of L-lactide and ϵ-caprolactone-poly(CL-LA) are bioabsorbable elastic materials and their clinical applications have been studied.[302,303]

10.1.2. *Bone fixation devices*

Although metal fixation in fracture treatment for undisturbed bone healing is a successful procedure, cortical bone and steel have very different mechanical properties. The elasticity constant of bone is only 1/10 that of implanted steel while tensile strength is 10 times lower.[304] Thus, the removal of metal implants can result in weakened bone with a danger of refracture. Biodegradable implants can meet the dynamic processes of bone healing, decreasing the weight-bearing of the material. After months, the entire material will disappear completely and no secondary surgery is required. PGA, PLA, polydioxanone and PHD have potential roles in this area. For clinical applications, polydioxanone was recommended for ligament augmentation, for securing a ligament suture, as a kind of internal splinting suture and as a kind of internal splinting to allow for early motion of the extremities after an operation.

Biodegradable polymers are useful for many other applications. A marrow spacer can help to save autologous bone material. A plug for closing the bone marrow is employed for endoprosthetic joint replacement. Fibres are used for filling large bone defects without mechanical loads.[304]

10.1.3. *Vascular grafts*

Many studies have been undertaken to develop acceptable small diameter vascular prostheses.[305–307] Among them, Niu *et al.*[305] designed small diameter vascular prostheses with incorporated matrices that can be absorbed into a growing anastomotic neointima. It was pointed out that a gelatin–heparin complex when adequately cross-linked, could simultaneously function as a temporary antithrombogenic surface and as an excellent substructure for an anastomotic neointima.

10.1.4. *Adhesion prevention*

Tissue adhesion after surgery occasionally causes serious complications. Materials that prevent tissue adhesion should be flexible and tough enough to provide for a tight cover over

the traumatized soft tissues, and should be biodegradable and reabsorbable after the injured tissue is completely regenerated. Matsuda *et al.*[308,309] developed photocurable mucopolysaccharides for a newly designed tissue adhesion prevention material that meets numerous requirements such as nonadherent surface characteristics, biocompatibility, biodegradability in accordance with the wound healing rate and nontoxicity.

Mucopolysaccharides (hyaluronic acid and chondronitin sulphate) partially functionalized with photoreactive groups, such as cinnamate or thymine, were subjected to UV irradiation to produce water-insoluble gels via intermolecular photodimerization of the photoreactive groups. Fig. 10[308] illustrates the preparation and photogelation of mucopolysaccharide gels. Photocured films with lower degrees of substitution, which had high water swellability and flexibility, prevented tissue adhesion and exhibited enhanced biodegradability. It was suggested that these newly designed mucopolysaccharide gels may aid injured tissue repair in a bioactive manner.

10.1.5. *Artificial skin*

For healing burns, skin substitutes or wound dressings made of biodegradable polymeric materials have recently been developed. Until now, most of the commercially developed artificial skins have utilized biodegradable polymers such as collagen,[310] chitin[311] and poly-L-leucine,[312,313] which are enzymatically degradable polymers.

Recently, Koide *et al.*[314] developed a new type of biomaterial for artificial skin, in the form of a sponge, by combining fibrillar collagen (F-collagen) with gelatin. The sponge was physically and metabolically stabilized by introducing crosslinks. Although several types of collagen-based artificial skins have been developed,[315–317] some undesirable characteristics of native collagen were noticed,[318] such as inducing rodlike shapes in fibroblasts and enhancing the expression of collagenase genes in fibroblasts. F-collagen with gelatin was found to overcome the above problems.

Yasutomi *et al.*[319] developed a biosynthetic wound dressing with a drug delivery capability. This medicated wound dressing is composed of a spongy mixture sheet of chitosan-derivatized collagen, which is laminated with a gentamycin sulphate impregnated polyurethane membrane. From *in vitro* evaluation, it was shown that this wound dressing is capable of suppressing bacterial growth and minimizing cellular damage. Evaluation of this wound dressing was conducted in 80 clinical cases including superficial second-degree burns, deep second-degree burns, donor sites and pressure sores, and achieved excellent results.

The development of hybrid artificial skins is also another important target in biomedical engineering. Here, synthetic polymers and cell cultures are combined to form a synthetic–biological composite. In this case, a biodegradable polymer may be required as the template for growing cells and tissue cultures *in vivo*.

10.1.6. *Drug delivery systems*

A new dimension for the use of polymeric materials as drug delivery devices involves incorporation of biodegradability into the system. A number of degradable polymers are potentially useful for this purpose, including a variety of synthetic and natural substances. The use of intentionally degradable polymers in medicine has been brought into prominence with new innovations in drug delivery systems. The limitations of conventional methods of

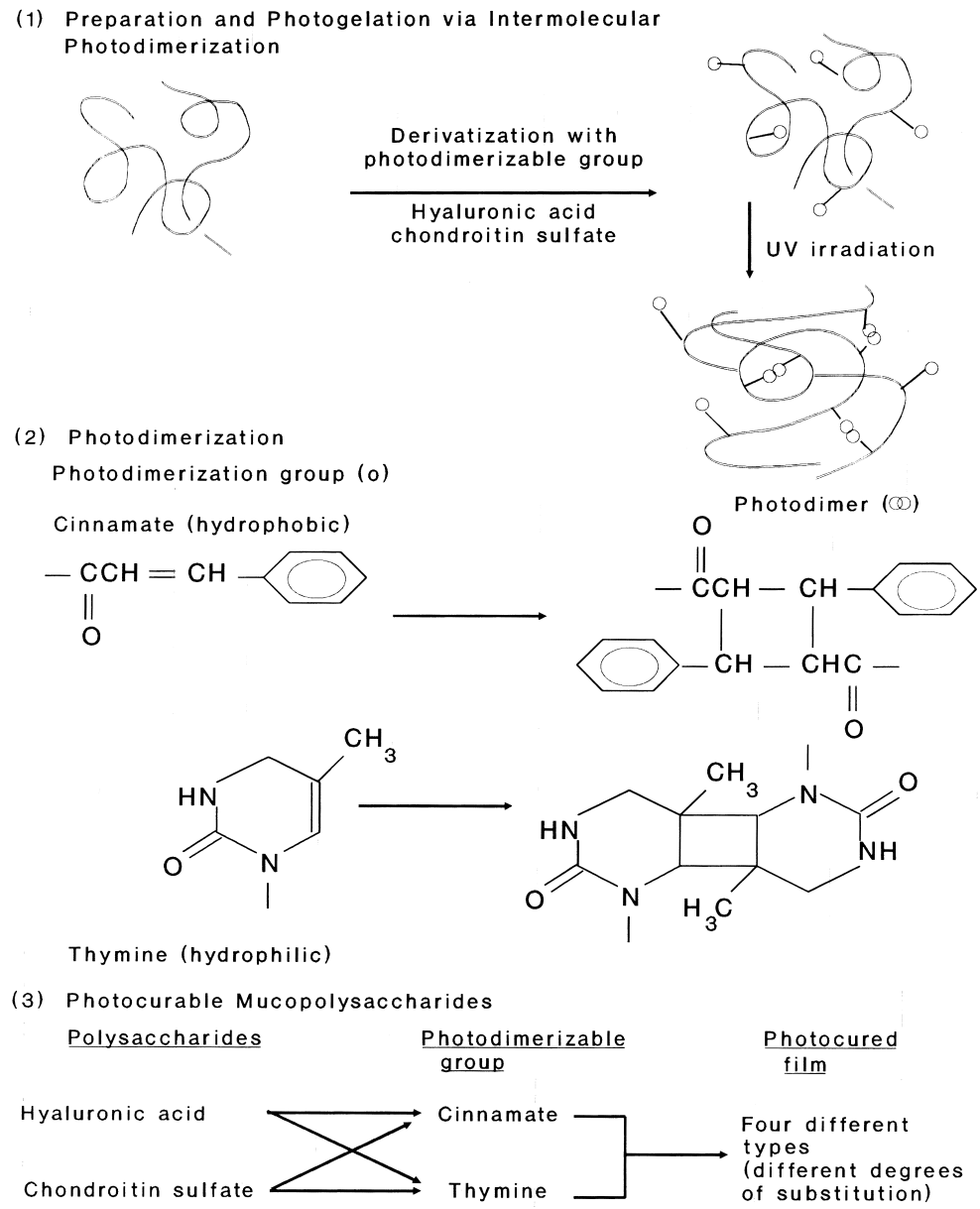


Fig. 10. Preparation and photogelation of mucosaccharides derivatized with cinnamate or thymine groups.

drug delivery, by tablet or injection for example, are well known. As a dose is applied, the plasma levels will be raised but these will be rapidly decreased as the drug is metabolized and will soon be below therapeutic levels. The next dose takes the plasma level high again and a cyclical pattern may be established, with most of the drug plasma levels possibly being

outside the optimal range. In addition, the drug usually permeates throughout the body and it is not targeted to the location where it is specifically required.

Amongst the many possible solutions to these problems is the use of controlled drug delivery systems,[320,321] from which the drug is released at a constant, predetermined rate, and possibly targeted to a particular site. One of the most prominent approaches is that in which the drug is contained within a polymer membrane or is otherwise encapsulated in a polymer matrix and where the drug diffuses out into the tissues following implantation. In some cases, erosion or dissolution of the polymer contributes to the release mechanism. Degradable polymers such as poly(lactic acid) and poly(ortho ester)s, are used for drug delivery systems.

Certain soluble polymers may be used as carriers for targeted drugs. Duncan and Kopacek[322] have reported the use of a variety of polymers to which are attached, via side groups, certain drugs which can be released following cleavage of the bonds attaching them to the backbone. The targeting or selectivity here is achieved by using bonds which are cleaved only under certain conditions, for example by liver enzymes, which permit the release of the drug only at their specific site of action.

The design of a plasticized, biodegradable polymeric material, suitable for application as a drug-delivery system, was attempted.[323] A poly(DL-lactic acid) oligomer was plasticized with 1,2-propylene glycol and glycerol. The latter plasticizer showed poor compatibility whereas 1,2-propylene glycol was compatible with the polymer up to high concentrations. The mixtures prepared displayed considerable depression of processing temperatures and enhanced deliveries of salicylic acid in the early stages of release. It seemed, therefore, feasible to produce systems which allow easy and safe processing and can be injected into a body cavity without the need for surgical retrieval after completion of the release. Furthermore, the differential rates of drug delivery might be of profound interest for cases where elevated drug doses are necessary in the beginning of treatment.

10.2. *Agricultural applications*

Since the introduction of plastic films in the 1930s and 1940s for greenhouse coverings, fumigation and mulching, agricultural applications of polymers have grown at an enormous rate. All principal classes of polymers, i.e. plastics, coatings, elastomers, fibres and water-soluble polymers are presently utilized in applications which include the controlled release of pesticides and nutrients, soil conditioning, seed coatings, gel plantings and plant protection. However, degradable plastics are also of interest as agricultural mulches and agricultural planting containers. Ultimate biodegradability, as in composting, is also of some interest as it would permit degradable plastics to be combined with other biodegradable materials and converted into useful soil-improving materials.

10.2.1. *Agricultural mulches*

Mulches permit growers to use plastic films to help with plant growth and then photodegrade in the fields thereby avoiding the cost of removal. The plastic films are desirable because they conserve moisture, reduce weeds and increase soil temperatures, thus improving the rate of growth in plants. For example, a 6 ha melon farm reported a two- to three-fold

increase in yield and ripening two weeks earlier as a result of using black polyethylene mulch. Elimination of weeds and avoidance of soil compaction by the use of mulch eliminates the need for cultivation, therefore root damage and stunting or killing of plants is further avoided. Fertilizer and water requirements are also reduced.[324]

Transparent polyethylene is more effective in trapping heat than black or smoke-grey films: soil temperatures may rise 5.5°C under clear films, as compared to 1.7–2.7°C under black films. Radiative heat loss at night, as the soil cools, is lessened by polymer films. In some cases, weed control has been reported because of solar heating of the polyethylene mulches.[325] If left in place, however, conventional films can cause problems during harvesting or during cultivating operations the next year. Removal and disposal are costly and inconvenient. Therefore, interest in the development of biodegradable or photodegradable films with short service lifetimes has grown. Although a large number of polymer types could be designed for controlled degradation, only a few have been commercialized.[85,326–330] Plastics used for this purpose usually contain light-sensitizing additives which cause the materials to undergo photodegradation.

The plastics used for mulch films are generally low density polyethylenes, poly(vinyl chloride), polybutylene or copolymers of ethylene with vinyl acetate. A particularly interesting photodegradable system consists of a mixture of ferric and nickel dibutyldithiocarbamates, the ratio of which is adjusted to provide protection for specific growing periods. The degradation is tuned so that when the growing season is over the plastic will begin to photodegrade. Another additive system being proposed for this application includes a combination of substituted benzophenones and titanium or zirconium chelates. The principal commercial degradable mulch is photodegradable poly(1-butene).[85]

Biodegradable films based on starch with poly(vinyl alcohol)[331] poly(ethylene-*co*-acrylic acid)[2,12] and poly(vinyl chloride)[332] have been developed in the USDA laboratories. Polylactone and poly(vinyl alcohol) films are readily degraded by soil microorganisms, whereas the addition of iron or calcium accelerated the breakdown of polyethylene.[333] Degradable mulches should break down into small brittle pieces which pass through harvesting machinery without difficulty and do not interfere with subsequent planting.

Effective fumigant mulches require reduced-porosity films which reduce the escape of volatile chemicals, i.e. nematocides, insecticides, herbicides, etc., and therefore allow for lower application rates.

10.2.2. *Controlled release of agricultural chemicals*

Controlled release (CR) is a method by which biologically active chemicals are made available to a target species at a specified rate and for a predetermined time. The polymer serves primarily to control the rate of delivery, mobility, and period of effectiveness of the chemical component. The principal advantage of CR formulations is that less chemicals are used for a given time period, thus lowering the impact on nontarget species and limiting leaching, volatilization, and degradation. The macromolecular nature of polymers is the key to limiting chemical losses by these processes.

CR polymeric systems can be divided into two broad categories. In the first, the active agent is dissolved, dispersed, or encapsulated by the polymeric matrix or coating. Release generally occurs by diffusion processes or by the biological or chemical breakdown of the matrix. In the second category polymers contain the active agent as part of the macromolecular

backbone or pendent side chain. Release results from biological or chemical cleavage of the bond between the bioactive agents and the polymer.

Physical systems that incorporate agricultural chemicals include microcapsules, physical blends, dispersions in plastics, laminates, hollow fibres and membranes. Kinetic models for release have been developed for each type of device.[334–338]

Starch,[339–345] cellulose[346,347] chitin,[335,346–348] alginic acid,[334] and lignin[334,349] are among the natural polymers used in CR systems. These have the advantages of being abundant, relatively inexpensive, and biodegradable. Although they possess functionality for derivatization, they have the one significant disadvantage of being insoluble in standard solvents suitable for encapsulation, dispersion, and formulation. Systems have been developed that overcome the solvent problem by *in situ* encapsulation, whereby gelantinized starch containing a chosen pesticide is cross-linked by adding calcium chloride[339] or boric acid,[340] or by xanthanation followed by oxidation.[341] The pesticide, as a result, is entrapped within the granular particles formed.

One of the largest applications for CR technology in agriculture is with fertilizers[350–352] Urea, a significant nitrogen source, easily reacts with formaldehyde to form a polymer. Subsequent hydrolysis of the polymer yields urea. Therefore, this is a simple and inexpensive system for CR.

10.2.3. *Agricultural planting containers*

A small niche for degradable plastics is the use of polycaprolactone for small agricultural planting containers. Although this is a small volume application for degradable plastics, it is presented here because it is one of the few applications in which the polymer used is biodegradable within a reasonable period of time. These polycaprolactone planting containers have been used for automated machine planting of tree seedlings. Within six months in the soil, the polycaprolactone was found to undergo significant biodegradation, resulting in 48% weight loss, with 95% weight loss occuring in a year.[78]

10.3. *Packaging*

Physical characteristics of packaging polymers are greatly influenced by the chemical structure, molecular weight, crystallinity and processing conditions of the polymers used. The physical characteristics required in packaging depend on what item will be packaged as well as the environment in which the package will be stored. Items which must be kept frozen for a period of time require special packaging. Food items require more stringent packaging requirements than nonperishable goods.

The challenge in the development of biodegradable packaging will be to combine polymers which are truly biodegradable into a laminate film or a film blend which has properties as good as those found in synthetic laminates. For food applications, for example, it may be possible to coat food items with pullulan which has a very low oxygen permeability and is edible and to utilize PHBV as an outer packaging which has good flexibility and is a moisture barrier. A film blend of pullulan and PHBV can also be produced[254] since both polymers can be melt blended under conditions where sufficient moisture is maintained during processing. The addition of pullulan to PHBV may reduce oxygen permeability and increase biodegradability of the blend due to the increased surface area of PHBV exposed following the rapid removal of pullulan due to its water solubility.

Several polysaccharide-based biopolymers are being used as possible coating materials or packaging films. They include starch, pullulan and chitosan. The degradation of synthetic polymer films can be accelerated by incorporating starch as a filler. LDPE blends with up to 10% corn starch were produced using conventional techniques and were made into bags for groceries or rubbish.

Pullulan is comprised primarily of maltotrise units connected by α-1,6 linkages. It is produced by several fungi as an extracellular secondary metabolite. Pullulan has been commercialized as a food source in Japan due to its natural origin and has been approved for food coatings.[353] It is a water-soluble polymer which produces clear, edible films which exhibit low oxygen permeability. Films can be produced by casting a 1–20% aqueous solution of pullulan on a metal plate roller. Pullulan, like starch, can also be moulded with heat and pressure if sufficient water is present as a plasticizer.[354]

A plant-derived polysaccharide is being investigated for film and coating applications. Konja flour is obtained from tubers of a perennial Amorphophallus herb species, cultivated in Asia and is comprised of approximately 1.6 mannose units to 1 glucose unit with β-1,4 linkages and randomly distributed acetyl side groups on the sugar molecules. The polysaccharide is obtained in the form of a flour called Nuticol® konja flour. Konja flour (1%) is stirred into a 60°C glycerin (1%)/water solution mixed in a blender for 15 min to reduce viscosity and then cast into a film.[355] Film properties can be altered by reacting the konja flour/glycerin mixture with 0.1% potassium carbonate or 0.2% calcium carbonate before film casting. Depending on the film preparation procedure, the films are transparent to opaque, gelatinous to strong in hot water and can exhibit low to high tear strengths. The physical and chemical properties of these films have not been quantified, but konja flour is generally recognized as safe (GRAS) as defined by the US Federal Food, Drug and Cosmetic Act, so it may have food coating or packaging applications in the future.

Poly-L-lactic acid (PLLA) is formed by a chemical condensation reaction of the lactic acid monomer and has a tensile strength at break of 45–70 MPa and an elongation of 85–105%. Argonne National Laboratory has patented a process to produce glucose from potato starch in 10 h instead of 100 h.[356] The glucose is then fermented to lactic acid and purified. The same laboratory estimates that the price for lactic acid produced from potato wastes is low enough to produce PLLA and manufacture biodegradable packaging items at a reasonable price. PLLA-based packagings under consideration include grocery and rubbish bags, diaper backings, six-pack rings and fast food containers.

REFERENCES

1. Fukuda, K., An overview of the activities of the Biodegradable Plastic Society, in *Biodegradable Polymers and Plastics*, ed. M. Vert *et al*. Royal Society of Chemistry, 1992, p. 169.
2. Otey, F. H., Westhoff, R. P. and Russell, C. R., *Ind. Eng. Chem. Prod. Res. Dev.*, 1977, **16,** 305.
3. Huang, J. C., Shetty, A. S. and Wang, M. S., *Adv. Polym. Technol.* , 1990, **10 (1),** 23.
4. Rutenberg, M. and Solarek, D., in *Starch: Chemistry and Technology*, ed. R. L. Whistler, J. Bemiller and E. Paschall. Academic Press, New York, 1984, p. 311.
5. Parandoosh, S. and Hudson, S., *J. Appl. Polym. Sci.*, 1993, **48,** 787.
6. Shulman, J. and Howarth, J. T., US Patent No. 3,137,664, 16 June 1964.
7. Griffin, G. J. L., *Am. Chem. Soc. Div. Org. Coat, Chem. Soc. Div. Org. Coat. Chem*, 1973, **33 (2),** 88.
8. Griffin, G. J. L. and Turner, R. D., *International Biodeterioration Conference*, Berlin, 1978.

9. Griffin, G. J. L., Advances in Chemistry Series, 134, *Fillers and Reinforcements for Plastics*. American Chemical Society, 1974, p. 159.
10. Otey, F. H., Mark, A. M., Mehltretter, C. L. and Russel, C. R., *Ind. Eng. Chem. Prod. Res. Dev.*, 1974, **13,** 90.
11. Otey, F. H. and Doane, W. M., A starch based degradable plastic film, in *Proc. Society of the Plastic Industry, Symp. on Degradable Plastics*, Washington, DC, June 1987, pp. 39–40.
12. Otey, F. H., Westhoff, R. P. and Doane, W. M., *Ind. Eng. Chem. Prod. Res. Dev.*, 1980, **19 (4),** 592.
13. Westhoff, P., Otey, F. H., Mehltretter, C. L. and Russell, C. R., *Ind. Eng. Chem. Res. Dev.*, 1974, **13,** 123.
14. Otey, F. H., Westhoff, R. P. and Russell, C. R., Starch based plastics and films, in *Proc. Tech. Symp. Nonwoven Product Technol*. International Nonwoven Disposables Association, Miami Beach, FL, March 1975.
15. Otey, F. H., Westhoff, R. P., Kwolek, W. F. and Mehletter, C. L., *Ind. Eng. Chem. Prod. Res. Dev.*, 1969, **8,** 267.
16. Otey, F. H., Westhoff, R. P. and Mehletter, C. L., *Staerke*, 1972, **24,** 267.
17. Boggs, W., US Patent No. 2,908,657, 13 October 1959.
18. Dosmann, P. and Steel, R. N., US Patent No. 3,004,934, 17 October 1961.
19. Bennett, L., Otey, F. H. and Mehltretter, C. L., *J. Cell. Past.*, 1967, **3,** 369.
20. Liungdahl, K. E. Erisksson, in *Advances in Microbial Ecology*, Vol. 8, ed. K. C. Marshall. Plenum, New York, 1985, p. 237.
21. Aubert, J. P., Beguin, P. and Millet, J., *Biochemistry and Genetics of Cellulose Degradation*. Academic, New York, 1988.
22. Gardener, R. M., Buchman, C. M., Komark, R., Dorschel, D., Boggs, C. and White, A. W., *J. Appl. Polym. Sci.*, 1994, **52 (10),** 1477.
23. Reese, E. T., *Ind. Eng. Chem.*, 1957, **49,** 89.
24. Cantor, P. A. and Mechalas, B. J., *J. Polym. Sci. Part C*, 1969, **28,** 225.
25. Gue, Ji-Dong, McCarthy, S. P., Smith, G. P., Eberiel, D. and Gross, R. A., *Proc. ACS Div. Polym. Mater. Sci. Eng.*, 1992, **67,** 230.
26. Buchanan, C. M., Gardner, R. M., Komarek, R. J., Gedon, S. C. and White, A. W., in *Fundamentals of Biodegradable Polymers and Materials*. Technomic, Lancaster, PA, 1993.
27. Buchanan, C. M., Gardner, R. M. and Komarek, R. J., *J. Appl. Polym. Sci.*, 1993, **47,** 709.
28. Komarck, R. J., Gardner, R. M., Buchanan, C. M. and Gedon, S., *J. Appl. Polym. Sci.*, 1993, **50,** 1739.
29. Stutzenberger, F. and Kahler, G., *J. Appl. Bacteriol.* , 1986, **61,** 225.
30. Chandy, T. and Sharma, C. P., *Biomat. Art. Cells Art. Org.*, 1990, **18,** 1.
31. Hosokawa, J., Nishiyama, M., Yoshihara, K. and Kubo, T., *Ind. Eng. Chem. Res.*, 1990, **29,** 800.
32. Muzzarelli, R. A. A., *Chitin in Nature and Technology*. Plenum Press, New York, 1986.
33. Muzzarelli, R. A. A., *Polym. Prepr. Am. Chem. Soc. Div. Polym. Chem.*, 1990, **31 (1),** 626.
34. Tokura, S., Miura, Y., Uraki, Y., Watanabe, K., Saiki, I. and Azuma, I., *Am. Chem. Soc. Div. Polym. Chem.*, 1990, **31 (1),** 627.
35. Tashibana, M., Yaita, A., Tamiura, H., Fukasawa, K., Nagasue, N. and Nakanura, T., *Jpn. J. Surg.*, 1988, **18,** 533.
36. Badwan, A. A., Abumalloh, A., Sallam, E., Abukalaf, A. and Jawan, O., *Drug Dev. Ind. Pharm.*, 1985, **11,** 239.
37. Yotsuyanagi, T., Ohkubo, T. and Ikada, K., *Chem. Pharm. Bull.*, 1987, **35,** 1555.
38. Pfister, G., Bahadir, M. and Korte, F., *J. Controlled Release*, 1986, **3,** 229.
39. Posillico, E. G., *Biotechnology*, 1986, **4,** 114.
40. King, G. A., Daugulis, A. J., Faulkner, P. and Goosen, M. F. A., *Biotechnol. Prog.*, 1987, **3,** 231.
41. Kwok, K. K., Groves, M. J. and Burgess, D. J., *Pharm. Res.*, 1991, **8,** 341.
42. Eastoe, J. E. and Leach, A. A., in *The science and Technology of Gelatin*, ed. A. G. Ward and A. Courts. Academic, New York, 1977, p. 73.
43. Tabata, Y. and Ikada, Y., *J. Pharm. Pharmacol.*, 1987, **39,** 698.
44. Tabata, Y. and Ikada, Y., *Pharm. Res.*, 1989, **6,** 422.

45. Tabata, Y., Uno, K., Muramatsu, S. and Ikada, Y., *Jpn. J. Cancer Res.*, 1989, **80,** 387.
46. Lt Yan, C. X. and Chen, X., *Biomaterials*, 1991, **12,** 640.
47. Tabata, Y., Uno, K., Ikada, Y. and Murametsu, S., *J. Pharm. Pharmacol.*, 1993, **45,** 303.
48. Tabata, Y., Uno, K., Ikada, Y., Kishida, T. and Muramatsu, S., *Jpn. J. Cancer Res.*, 1993, **84,** 681.
49. Tabata, Y. and Ikada, Y., *J. Controlled Release*, 1993, **27,** 79.
50. Kamath, K. R. and Park, K., *Proc. Int. Symp. Control. Release Bioact. Mater.*, 1992, **19,** 42.
51. Ziegler, G. R., *Biotechnol. Prog.*, 1991, **7,** 283.
52. Shinde, B. G., Nithianandam, V. S., Kaleem, K. and Erhan, S., *Bio-Medical Mater. Eng.*, 1992, **2,** 123.
53. Shinde, B. G. and Ehran, S., *Bio-Medical Mater. Eng.*, 1992, **2,** 127.
54. Kuwajima, T., Yoshida, H. and Hayashi, K., *J. Appl. Polym. Sci.*, 1976, **20,** 967.
55. Kumar, G. S., Kaplagam, V. and Nandi, U. S., *J. Appl. Polym. Sci.*, 1981, **26,** 3633.
56. Doi, Y., *Microbial Polyester*. VCH, New York, 1990.
57. Anderson, A. J. and Dawes, E. A., *Microbial Rev.*, 1990, **54,** 450.
58. Brandl, H., Gross, R. A., Lenz, R. W. and Fuller, R. C., *Adv. Biochem Eng. Biotech.*, 1990, **41,** 77.
59. Marchessault, R. H., Blulim, T. L., Destandes, Y., Hamer, G. K., Orts, W. J., Gundararajan, P. R., Taylor, M. G., Bloembergen, S. and Holden, D. A., *Makromol Chem. Makromol. Symp*., 1988, **19**, 235; Owen, A. J., Bauer, H. and Owen, A. J., *Colloid Polymer Sci*., 1988, **226**, 241.
60. Holms, P. A., Wright, L. F. and Collins, S. H., Eur. Patent No. 0,052,459, 1982; Eur. Patent No. 0,069,497, 1983.
61. Preusting, H., Nijenhuis, A. and Witholt, B., *Macromolecules*, 1990, **23,** 4220.
62. Bloembergen, S., Holden, D. A., Bluhm, T. L., Hamer, G. K. and Marchessault, R. H., *Macromolecules*, 1989, **22,** 1656.
63. Bloembergen, S., Holden, D. A., Bluhm, T. L., Hamer, G. K. and Marchessault, *Macromolecules*, 1989, **22,** 1663.
64. Doi, Y. and Abe, C., *Macromolecules*, 1990, **23,** 3705.
65. Kunioka, M., Kawaguchi, Y. and Doi, Y., *Appl. Microbiol. Biotechnol.*, 1989, **30,** 569.
66. Kunioka, M., Tamaki, A. and Doi, Y., *Macromolecules*, 1989, **22,** 694.
67. Kunioka, M., Nakamura, Y. and Doi, Y., *Polym. Commun.*, 1988, **29,** 174.
68. Doi, Y., Tamaki, A., Knuioka, M. and Soga, K., *J. Chem. Soc. Chem. Commun*., 1987, 1635.
69. Doi, Y., Knuioka, M. and Nakamura, Y., *Macromolecules*, 1987, **20,** 2988.
70. Doi, Y., Tamaki, A., Knuioka, M. and Nakamura, Y., *J. Chem. Soc. Macromolecules*, 1986, **19,** 2860.
71. Holland, S. J., Yasin, M. and Tighe, B. J., *Biomaterials*, 1990, **11,** 206.
72. Doi, Y., Kanesawa, Y., Kawaguchi, Y. and Knuioka, M., *Makromol. Chem. Rapid Commun.*, 1989, **10,** 227.
73. Bhalakia, S. N., Patel, T., Gross, R. A. and McCarthy, S. P., *Polym. Prepr. Am. Chem. Soc. Div. Polym. Chem.*, 1990, **31 (1),** 441.
74. Dave, P. B., Ashar, N. J., Gross, R. A. and McCarthy, S. P., *Polym. Prepr. Am. Chem. Soc. Div. Polym. Chem.*, 1990, **31 (1),** 442.
75. Marchessault, R. H., Monasterios, C., Ramsay, B. and Saracovan, I., in *33rd IUPAC, Int. Symp. on Macromolecules*. IUPAC, Montreal, 1990, p. 264.
76. Fields, R. D. and Rodriguez, F., *Proc. 3rd Int. Biodegradation Symp*. Applied Science, New York, 1976, p. 775.
77. Bitritto, M. M., Bell, J. P., Brenchle, G. M., Huang, S. J. and Knox, J. R., *J. Appl. Polym. Sci. Appl. Polym. Symp.*, 1979, **35,** 405.
78. Potts, J. E., Cleudinning, R. A., Ackart, W. B. and Niegich, W. D., *Polymer and Ecological Problems*. Plenum Press, New York, 1973, p. 61.
79. Tokiwa, Y. and Suzuki, T., *J. Appl. Polym. Sci.*, 1981, **26,** 441.
80. Tokiwa, Y. and Suzuki, T., *Agric. Biol. Chem.*, 1971, **54,** 603.
81. Fiazza, E. J. and Schmitt, E. E., *J. Biomed. Mater. Res. Symp.*, 1971, **1,** 43.
82. Schmitt, E. E. and Polistina, R. A., US Patent No. 3 297 033.
83. Schmitt, E. E., Epstein, M. and Polistina, R. A., US Patent No. 3 422 871.
84. Glick, A. and McPherson, J. B., US Patent No. 3 626 948, 1981.

85. Potts J. E., in *Kirk-Othmer Encyclopeida of Chemical Technology*, Suppl. Vol., ed. M. Grayson. Wiley-Interscience, New York, 1984, p. 626.
86. Fields, R. D., Rodriquez, F. and Finn, R. K., *J. Appl. Polym. Sci.*, 1974, **18,** 3571.
87. Benedict, C. V., Cook, C. V., Jarrett, P., Cameron, J. A., Huang, S. J. and Bell, P., *J. Appl. Polym. Sci.*, 1983, **28,** 327.
88. Benedict, C. V., Cameron, J. A. and Huang, S. J., *J. Appl. Polym. Sci.*, 1983, **28,** 335.
89. Cook, W. J., Cameron, J. A., Bell, J. P. and Huang, S. J., *J. Polym. Sci., Polym. Lett.*, 1981, **19,** 159.
90. Jar, P., Huang, S. J., Bell, J. P., Cameron, J. A. and Benedict, C., *Org. Coat. Appl. Polym. Sci. Proc.*, 1982, **47,** 45.
91. Wasserman, D., US Patent No. 3,375,008.
92. Jarret, P., Cook, W. J., Bell, J. P., Huang, S. J. and Cameron, J. A., *Polym. Am. Chem. Soc. Div. Polym. Chem.*, 1981, **22,** 381.
93. Pitt, C. G., Marks, T. A. and Schindler, A., in *Controller Release of Bioactive Materials*, ed. R. Baker. Academic Press, New York, 1980, p. 9.
94. Jaffe, H., Giang, P. A., Hayes, D. K., Miller, J. A. and Stroud, B. H., *Controlled Release Pestiudes and Pharmaceuticals*. Plenum, New York, 1981, p. 303.
95. Pitt, C. G., Gratzl, M. M., Jeffcoat, A. R., Zweidinger, R. and Schindler, A., *Pharm. Sci.*, 1979, **68,** 1534.
96. Pitt, C. G., Chasaldo, F. J., Hibionada, J. M., Klimas, D. M. and Schindler, A., *Appl. Polym. Sci.*, 1981, **28,** 3779.
97. Clendinning, R. A., Potts, J. E. and Niegisch, W. D., (to Union Carbide Corp.), US Patent Nos. 3,850,862; 3,850,863 and 3,844,984, 1974.
98. Tokiwa, Y. and Suzuki, T., *Nature*, 1977, **270,** 76.
99. Tokiwa, Y. and Suzuki, T., *J. Fermen. Technol.*, 1974, **52,** 393.
100. Fields, R. D. and Rodriquez, F. (Eds), *Proc. 3rd Int. Biodegradation Symp.* Applied Science Publishers, New York, 1976, p. 775.
101. Tokiwa, Y. and Suzuki, T., *Agric. Biol. Chem.*, 1977, **41,** 265.
102. Tokiwa, Y., Ando, T. and Suzuki, T., *J. Ferment. Technol.*, 1976, **54,** 603.
103. Negoro, S., Taniguchi, T., Kanaoka, M., Kimura, H. and Okada, H., *J. Bacteriol.*, 1983, **155,** 22.
104. Kinoshita, S., Negora, S., Muramatsu, M., Bisaria, V. S., Sawada, S. and Okada, *Eur. J. Biochem.*, 1977, **80,** 489.
105. Kinoshita, S., Terada, T., Taniguchi, L. T., Takene, Y., Musuda, S., Matsunaga, N. and Okada, H., *Eur. J. Biochem.*, 1981, **116,** 547.
106. Kinoshita, S., Kageyama, S., Iba, K., Yamada, Y. and Okada, H., *Agric. Biol. Chem.*, 1975, **39,** 1219.
107. Fukumura, T., *J. Biochem.*, 1966, **59,** 537.
108. Watanabe, T. and Gakkaishi, S., 43, 192 (1987) (Chem. Abstr. 1987, 106, 215 390).
109. Huang, S. J., Bitritto, M., Leong, K. W., Paulisko, J., Roby, M. and Knox, J. R., *Adv. Chem. Ser.*, 1978, **169,** 205.
110. Bailey, W. J., Okamota, Y., Kuo, W. C. and Varita, T., *Proc. 3rd Int. Biodegradation Symp.* Applied Science, Barking, 1976, p. 765.
111. Tikiwa, Y., Suzuki, T. and Ando, T., *J. Appl. Polym. Sci.*, 1979, **24,** 1701.
112. Katakai, R. and Goodman, M., *Macromolecules*, 1982, **15,** 25.
113. Krichederf, H. R. and Jonte, J. M., *Polym. Bull.*, 1983, **9,** 276.
114. Barrows, T. H., Johnson, J. D., Gibson, S. J. and Girassing, D. M., *ACS Polym. Sci. Technol. Ser. Adv. Biomed. Polym.*, 1986, **34,** 85.
115. Carby, R. T. and Kaplan, A. M., *Appl. Microbiol.*, 1968, **16,** 900.
116. Haung, S. J., Macri, C., Roby, M., Benedict, C. and Cameron, J. A., *ACS Symp. Ser.*, 1981, **172,** 471.
117. Leong, K. W., Brott, B. C. and Langer, R., *J. Biomed. Mater. Res.*, 1984, **19,** 941.
118. Conix, A., *J. Polym. Sci.*, 1958, **29,** 343.
119. Langer, K. W., Amore, P. D., Marletta, M. and Langer, R., *J. Biomed. Mater. Res.*, 1986, **20,** 51.
120. Ron, E., Mathiowitz, E., Mathiowit, G. and Langer, R., *Polym. Prepr. Am. Chem. Soc. Div. Polym. Chem.*, 1989, **30 (1),** 462.

121. Albertsson, A. C. and Lundmark, S. J., *Macromol. Sci. Chem.*, 1988, **A 25,** 247.
122. Albertsson, A. C. and lundmark, S., *J. Macromol. Sci. Chem.*, 1990, **A 27,** 397.
123. Albertsson, A. C. and Lundmark, S., *Br. Polym. J.*, 1990, **23,** 205.
124. Heller, J. and Baker, R. W., in *Controlled Release of Bioactive Materials*, ed. R. Baker. Academic Press, New York, 1980, p. 1.
125. Hopfeukerg, H. B., in *AICNE Sym. Ser*. Vol. 77, ed. S. K. Chandrasekaran. 1981, p. 37.
126. Huang, S. J., Pavlikso, J. A., Benicewicz, B. and Wringer, E., *Polym. Repr. Am. Chem. Sco. Div. Polym. Chem.*, 1981, **22,** 56.
127. Casey, J. P. and Manly, D. C. (eds), *Proc. 3rd Int. Biodegradation Symp*. Applied Science Publishers, New York, 1976, p. 819.
128. Suzuki, T., *J. Appl. Polym. Sci. Appl. Polym. Symp.*, 1979, **35,** 431.
129. Watanabe, Y., Morita, M., Hamade, N. and Tsujisake, Y., *Agric. Biol. Chem.*, 1975, **39,** 2447.
130. Morita, M. and Watanabe, Y., *Agric. Biol. Chem.*, 1977, **41,** 1535.
131. Watanabe, Y., Hamada, N., Morita, M. and Tsuisake, J., *Arch. Biochem. Biophys.*, 1976, **174,** 575.
132. Fukanage, F., Sumina, S., Neda, K., Takemasa, T. and Tachibana, K., *Jpn. kokai*, 1977, 7,794,471.
133. Fukunaga, F., Nesta, K., Tuchibana, K., Akemara, T. T. and Sumina, S., *Jpn. kokai*, 1976, 76,25,786.
134. Huang, S. J., Wang, I. F. and Quinga, E., in *Modification of Polymers*, ed. C. F. Carraher and J. A. Moore. Plenum, New York, 1983, p. 75.
135. Huang, S. J., Quinga, E. and Wang, I. F., *Org. Coat. Appl. Polym. Sci. Proc.*, 1982, **46,** 345.
136. Huang, S. J. and Kitchen, O., The University of Connecticut, Storrs, CT, unpublished results; Kitchen, O. PhD dissertation, University of Connecticut, Storrs, CT, 1984.
137. Rosato, D. V., in *Environmental Effects on Polymeric Materials*, ed. D. V. Rosato and R. T. Schwartz. Wiley-Interscience, New York, 1968, p. 991.
138. Brown, A. E., *Mod. Plast.*, 1946, **23,** 189.
139. Darby, R. T. and Kaplan, A. M., *Appl. Microbiol.*, 1968, **6,** 900.
140. Shiev, W. T., in *Drug Carrier in Biology and Medicine*, ed. G. Gregoriadis. Academic Press, New York, 1979, p. 43.
141. Harris, F. W. and Post, L. K., *J. Polm. Sci. Polym. Lett. Ed.*, 1975, **13,** 225.
142. Harris, F. W. and Post, L. K., *Polym. Prepr. Am. Chem. Soc. Div. Polym. Chem.*, 1975, **16,** 622.
143. Barim, B. and Dcanin, R. D., *Polym. Plast. Technol. Eng.*, 1973, **2,** 1.
144. Hegyeli, A. F., *J. Biomed. Mater. Res.*, 1973, **7,** 205.
145. Couveur, P., Roland, M. and Speisen, P., US Patent No. 4,329,332, 1982.
146. Vezin, W. R. and Florence, T., *J. Biomed. Mater. Res.*, 1980, **14,** 93.
147. Leonard, F., Kulkarni, R. K., Brandes, G., Nelson, J. and Cameron, J. J., *Appl. Polym. Sci.*, 1960, **10,** 259.
148. Wade, W. R. and Leonard, F., *J. Biomed. Mater. Res.*, 1972, **6,** 25.
149. Leonard, F., *Org. Coat. Plast. Chem.*, 1971, **31,** 225.
150. Wichterle, O. and Lim, D., *Nature*, 1960, **185,** 117.
151. Andrade, J. D. (ed.), *ACS Symp. Ser*., 1976, **31**.
152. Kopecek, J. and Sprinel, L., *Polym. Medicine*, 1974, **4,** 109.
153. Timmer, W., *Chem. Technol.*, 1979, **9,** 175.
154. Refojo, M. F., in *Kirk-Othmer Encyclopedia of Chemical Technology*, 3rd edn, Vol. 6, ed. M. Grayson. Wiley-Interscience, New York, 1979, p. 720.
155. Sevcik, S., Stamburg, J. and Schmidt, P., *J. Polym. Sci., Part C*, 1967, **16,** 821.
156. Huang, S. J. and Wang, I. F., University of Connecticut, Storrs, CT, unpublished results; Wang, I. F., PhD dissertation, University of Connecticut, Storrs, CT, 1984.
157. Jatkewitz, V. H. and Naturfoursch, Z., *Teil*, 1955, **B 10,** 27.
158. Bartulin, J., Przybyski, M., Ringsdorf, H. and Ritter, H., *Makromol. Chem.*, 1974, **175,** 1007.
159. Batz, H. G., Ringsdorf, H. and Ritter, H., *Makromol. Chem.*, 1974, **175,** 2229.
160. Huang, S. J., Bansleben, D. A. and Knox, J. R., *J. Appl. Polym. Sci.*, 1979, **23,** 429.
161. Bailey, W. J., Okamoto, Y., Kuo, W. C. and Narita, T., in *Proc. 3rd Int. Biodegradation Symp.*, ed. J. M. Sharpley and A. M. Kaplan. Applied Science Publishers, New York, 1976, p. 765.

162. Varnell, W. D., Harnson, I. R. and Kosnioski, S. J., *J. Polym. Sci., Polym. Phys. Ed.*, 1981, **10,** 1237.
163. Blundell, D. J., Keller, A. and Ward, I. M., *J. Polym. Sci., Polym. Phys. Ed.*, 1966, **4,** 781.
164. Khambatta, F. B., Warner, F., Russell, T. and Stein, R. S., *J. Polym. Sci., Polym. Phys. Ed.*, 1976, **14,** 1391.
165. Patel, G. N. and Keller, A., *J. Polym Sci. Polym. Phys. Ed.*, 1975, **13,** 323.
166. Duong, D. T. and Bell, J. P., *J. Polym. Sci., Polym. Phys. Ed.*, 1975, **13,** 765.
167. Mandels, M. and Reese, E. T., *J. Bacteriol.*, 1957, **73,** 269.
168. Rosenberg, E., Zuckerberg, A., Rubinovitz, C. and Gutnik, D. L., *Appl. Environ. Microbiol.*, 1979, **37,** 402.
169. Blundell, D. J., Keller, A., Ward, I. M. and Grant, I. J., *J. Polym. Sci., Part A2*, 1968, **6,** 1621.
170. Udagawa, Y., *J. Polym. Sci., Part A2*, 1971, **9,** 1807.
171. Zeikov, G. E. and Livshitz, U. S., *Polym. Degrad. Stab.*, 1987, **17,** 65.
172. Huang, S. J., Bryne, C. and Palisko, J. A., *ACS Symp. Ser.*, 1980, **121,** 299.
173. Albertsson, A. C., *Preprints of the Int. Symp. on Characterization and Analysis of Polymers*, 1985, p. 477.
174. Albertsson, A. C., Anderson, S. O. and Karlsson, S., *Polym. Degrad. Stab.*, 1987, **18,** 73.
175. Williams, D. F., Chu, C. C. and Dwyer, J., *J. Appl. Polym. Sci.*, 1984, **29,** 1865.
176. Chu, C. C., *Polymer*, 1985, **26,** 591.
177. Walker, J. D., Austin, J. F. and Colwell, R. R., *J. Gen. Appl. Microbiol.*, 1975, **21,** 27.
178. Dwyer, D. F. and Tiegje, J. M., *Appl. Environ. Microbiol.*, 1986, **52,** 852.
179. Swisher, R. D., *Surfactant Biodegradation*. Marcel Dekker, 1987, p. 415.
180. *Proc. of Society of the Plastics Industry Symp. on Degradable Plastics*, Washington, DC, June 1987.
181. Seal, K. J., in *Chemistry and Technology of Biodegradable Polymers*, ed. G. J. L. Griffin. Blackie Academic Professional, 1992, p. 122.
182. Muller, R. J., Augusta, J. and Pantke, M., *Mater. Org.* , 1992, **27 (3),** 179.
183. *OECD Guidelines for Testing Chemicals*, OECD, Paris, 1981.
184. Seal, K. J. and Pantke, M., *Mater. Org.* , 1986, **21 (2),** 151.
185. Cole, M. A., *Preprints of Proc. ACS Division of Polymeric Materials Science and Engineering*, 1990, **63**, 877.
186. Bailey, W. J., Kuruganti, V. K. and Angle, J. S., *Macromolecules*, 1990, **23,** 149.
187. Bailey, W. J. and Gapud, B., *Polym. Stab. Deg.*, 1985, **280,** 423.
188. Scott, G., Mellar, D. C. and Moir, A. B., *Eur. Polym. J*., 1973, **9**, 219; Colin, G., Cooney, J. D., Carlsson, D. J. and Wiles, D. M., *J. Appl. Polym. Sci*., 1981, **26**, 509.
189. Guillet, J. E. (ed.), *Degradable Materials Perspective Issues and Opportunities*. CRC, Boca Raton, 1990, p. 55.
190. Scott, G., *Polymer News*, 1989, **14,** 169.
191. Gage, P., *TAPPI J*., 1990, 161.
192. Griffin, G. J. L., US Patent No. 4,016,117, 1977. Patent No. 1,487,050, 1977.
193. Otey, F. H., Westhoff, R. P. and Doane, W. M., *Ind. Eng. Chem. Res.*, 1987, **26,** 1659.
194. Roper, H. and Koch, H., *Starch/Starke*, 1991, **42,** 123.
195. Peanasky, J. S., Long, J. M. and Wool, R. P., *J. Polym. Sci., Part B: Polym. Phys.*, 1991, **29,** 565.
196. Goheen, S. M. and Wool, R. P., *J. Appl. Polym. Sci.*, 1991, **42,** 2691.
197. Koleske, J., in *Polymers Blends*, Vol. 2, ed. D. R. Paul and S. Newman. Academic, New York, 1978, p. 369.
198. Juni, K., Nakano, M. and Kuboto, M., *J. Controlled Release*, 1986, **4,** 25.
199. Koenig, W., Seidel, H. R. and Sandow, J. K., European Patent Appl. 133,988 AZ, 1985.
200. Dave, P., Ashar, N., Gross, R. and McCarthy, S. P., *Polym. Preprints*, 1990, **33,** 1.
201. Agarwal, A., Singh, A. and Agarwal, P. N., *Pop. Plast.*, 1971, **16 (Oct.)**.
202. Griffin, G. J. L., *J. Polym. Sci.*, 1976, **57,** 281.
203. Severini, F., Gallo, R. and Ipsali, S., *Polym. Deg. Stab.*, 1987, **17,** 57.
204. Severini, F., Gallo, R. and Ipsali, S., *Polym. Deg. Stab.*, 1986, **14,** 341.
205. Albertsson, A. C. and Ranby, B., in *Proc. 3rd Int. Biodegradation Symp.*, ed. J. M. Sharpley and A. M. Kaplan. Applied Science Publishers, London, 1976, p. 743.

206. Corbin, G. and Henman, J. J., in *Proc. 3rd Int. Conf. on Advances in the Stabilization of Polymers*, Lucerne, Switzerland, June 1981, p. 116.
207. Albertsson, A. C., *J. Appl. Polym. Sci.*, 1980, **25,** 1655.
208. Colin, G., Cooney, J. D., Carlsson, D. J. and Wiles, D. M., *J. Appl. Polym. Sci.*, 1981, **26,** 509.
209. Albertsson, A. C. and Karlsson, S., *J. Appl. Polym. Sci.*, 1988, **35,** 1289.
210. Karlsson, S., Ljungquist, O. and Albertsson, A. C., *Polym. Degrad. Stab.*, 1988, **21,** 237.
211. Raghavan, D., *Polym. Eng. Sci.*, 1992, **32,** 6.
212. Siu, R. G. H., *Microbial Decomposition of Cellulose*. Reinhold, New York, 1951.
213. Lollar, R. M., in *Deterioration of Materials*, Chap. 8, ed. G. A. Greathouse and J. Wessel. Reinhold, New York, 1954.
214. Griffin, G. J. L., *Pure Appl. Chem.*, 1980, **52,** 399.
215. Barua, P. K., *Appl. Microbiol.*, 1970, **20,** 657.
216. Merdinger, E. and Merdinger, R. P., *Appl. Microbiol.*, 1970, **20,** 651.
217. Miller, T. C. and Johnson, M. J., *Biotech.*, 1966, **8,** 567.
218. Sokatch, J. R., *Bacterial Physiology and Metabolism*. Academic Press, London, 1969, p. 157.
219. Foster, J. W. and Leeuwenhock, V. J., *Microbiol. Serol.*, 1962, **28,** 241.
220. McKenna, E. J. and Kallio, R. E., *Am. Res. Microbiol.*, 1965, **19,** 183.
221. Vander Linder, A. C. and Thysee, C. J. E., *Adv. Enzymol.*, 1965, **27,** 469.
222. Stewart, J. E., Kallio, R. E., Stevenson, D. P., Jones, A. C. and Schissler, D. O., *Bacteriol.*, 1959, **78,** 441.
223. Leadbetter, E. R. and Foster, J. W., *Arch. Biochem. Biophys.*, 1959, **32,** 491.
224. Lukins, H. B. and Foster, J. W., *J. Bacteriol.*, 1963, **85,** 1074.
225. Potts, J. E., Clendinning, R. A. and Ackart, W. B., USEPA Contract No. CPE-70-124, 1972, p. 29.
226. Nykvist, N. B., *Plastics and Polymers*, 1975, **195 (Oct.)**.
227. Potts, J. E., Clendining, R. A., Ackartand, W. B. and Niegisch, W. D., *Tech. Pap. Reg. Tech. Conf. Soc. Plant Eng.*, 1972, **63 (Oct. 11-12)**.
228. Kestelman, B. N., Yarovenko, V. L. and Melnikova, E. J., *Int. Biodet. Bull.*, 1972, **8,** 15.
229. Heap, W. M. and Morrell, S. H., *J. Appl. Chem.*, 1968, **18,** 189.
230. Dolezel, B., *Brit. Plast.*, 1967, **40,** 105.
231. Demmer, F., *Mater. Org.*, 1968, **3,** 19.
232. Kuster, E. and Azadi-Bakhsh, A., in *Proc. of Degradability of Polymers and Plastics Conf.* Institute of Electrical Engineers, London, 1973, p. 16/1.
233. Wallhauser, K. H., in *Proc. of Degradability of Polymers and Plastics Conf.* Institute of Electrical Engineers, London, 1973, p. 17/1.
234. Griffin, G. J. L., Preprints, *15th Prague Microsymposium on Degradation and Stabilization of Polyolefins*, Prague, July 1974.
235. Albertsson, A. C., *Eur. Polym. J.*, 1980, **16 (7),** 623.
236. Albertsson, A. C., *J. Appl. Polym. Sci.*, 1978, **22,** 3419.
237. Albertsson, A. C., Banhidi, Z. G. and Ericsson, L. L. B., *J. Appl. Polym. Sci.*, 1978, **22,** 3435.
238. Schwartz, A., *Mikrobielle Korrosion Von Kunst Stoffen and Ihren Bestandteilen*. Akademic-Verlag, Berlin, 1963.
239. Heap, W. M. and Morrell, S. H., *J. Appl. Chem.*, 1968, **18,** 189.
240. Rodriguez, F., *Chem. Technol*., 1971, 409.
241. Hueck, H. J., *Int. Biodeter. Bull.*, 1974, **10,** 87.
242. Connolly, R. A., in *Biodeterioration of Materials*, Vol. 2, ed. A. H. Walters and E. H. Hueck. Applied Science, London, 1972, p. 168.
243. Bredley, S. A. and Carr, S. H., *J. Appl. Phys.*, 1973, **44,** 4296.
244. Suzuki, T., Ichihara, Y., Yamada, M. and Tonomura, K., *Agric. Biol. Chem.*, 1973, **36,** 747.
245. Potts, J. E., Clendinning, R. A., Ackart, W. E. and Niegisch, W. D., *Polym. Prep. Am. Chem. Soc. Div. Polym. Chem.*, 1970, **13,** 629.
246. Baum, B. and Deanin, R. D., *Polym. Plast. Technol. Eng.*, 1973, **2,** 1.
247. Gorzynaska, J., *Int. Biodet. Bull.*, 1973, **10 (3),** 87.
248. Otey, F. H., Westhoff, R. P. and Mehltretter, C. L., *Starke*, 1972, **24 (4),** 107.
249. Albertsson, A. C. and Ranby, B., *J. Appl. Polym. Sci., Appl. Polym. Symp.*, 1978, **35,** 423.

250. Otey, F. H., Mark, A. M., Mehltretter, C. L. and Russell, C. R., *Ind. Eng. Chem. Prod. Res. Dev.*, 1974, **13,** 90.
251. Geetha, R., Torikai, A., Nagaya, S. and Fucki, K., *Polym. Degrad. Stab.*, 1987, **19,** 279.
252. Weiland, M., Daro, A. and David, C., *Polym. Degrad. Stab.*, 1995, **48,** 275.
253. Guiller, J. E., Heskins, M. and Speoer, L. R., *J. Appl. Polym. Sci.*, 1988, **58,** 80.
254. Gage, P., *Tappi J.*, 1990, **73 (10),** 161.
255. Scott, G., *J. Photochem. Photobiol. Chem.*, 1990, **51,** 73.
256. Swift, G., in *Agricultural and Synthetic Polymers Biodegradability and Utilization*, ACS Symposuim Series 433, ed. J. E. Glass and G. Swift. ACS, Washington, 1992, p. 2.
257. Albertsson, A. C. and Karlsson, S., in *Agricultural and Synthetic Polymers Biodegradability and Utilization*, ACS Symposuim Series 433, ed. J. E. Glass and G. Swift. ACS, Washington, 1990, p. 60.
258. Griffin, G. J. L., *Polym. Degrad. Stab.*, 1994, **45 (2),** 241.
259. Shogren, R. L. and Jasberg, B. K., *J. Environ. Polym. Degrad.*, 1994, **2 (2),** 99.
260. Raghavan, D., Wagner, G. C. and Wool, R. P., *J. Environ. Polym. Degrad.*, 1993, **1 (3),** 203.
261. Griffin, G. J. L. and Nathan, P. S., *J. Appl. Polym. Sci.*, 1979, **35,** 475.
262. Griffin, G. J. L., in *Proc. Symp. on Degradable Plastics*. Society of the Plastics Industry, Washington DC, 1987, p. 47.
263. McCarthy, S. P., Lisuardi, A. and Gada, R. A., in *Proc. Unique Tech. Conf. on Tomorrow's Materials Today*. Society of Plastics Engineering, Brookfield, CT, 1993, p. 311.
264. Otey, F. H. and Westhoff, R. P., *Ind. Eng. Chem. Prod. Res. Dev.*, 1984, **23,** 284.
265. Gould, J. M., Imam, S. H. and Gordon, S. H., in *Proc. ACS Division of Polymeric Materials, Science and Engineering*, Vol. 63. ACS, Washington, DC, 1990, p. 853.
266. Shogren, R. L., Gordon, S. H., Harry-O'Kuru, R. E. and Gould, J. M., *Proc. ACS Division of Polymeric Materials Science and Engineering*, Vol. 63. ACS, Washington, DC, 1990, p. 741.
267. Shogren, R. I., Thompson, A. R., Felker, F. C., Harry-O'Kuru, R. E., Gordon, S. H., Greene, R. V. and Gould, J. M., *J. Appl. Polym. Sci.*, 1992, **44 (11),** 1971.
268. Gonsalves, K. E., Patel, S. H. and Chen, X., *Annual Tech. Conf.* Society of Plastics Engineers, Brookfield Center, CT, 1989, p. 1356.
269. Swanson, C. L., Westhoff, R. P., Doane, W. M. and Otey, F. H., *Div. Polym. Chem*., 1987, **28**(2), 105.
270. Hueck, H. J., *Int. Biodet. Bull.*, 1994, **10 (3),** 87.
271. Gorzynska, J., *Przem. Ferment. Polny.*, 1973, **17 (5),** 36.
272. Albertsson, A. C., Griffin, G. J. L., Karlsson, S., Nishimoto, K. and Watanabe, Y., *Polym. Degrad. Stab.*, 1994, **45,** 173.
273. Breslin, V. T. and Li Boen, *J. Appl. Polym. Sci.*, 1993, **48,** 2063.
274. Peanasky, J. S., Long, J. M. and Wool, R. P., *J. Polym. Sci., Part B: Polym. Phys.* , 1991, **29,** 565.
275. Willett, J. L., *J. Appl. Polym. Sci.*, 1994, **54,** 1685.
276. Tsao, R., Anderson, T. A. and Coats, J. R., *J. Environ. Polym. Degrad.*, 1993, **1 (4),** 301.
277. Posey, T. and Hester, R. D., *Plastics Eng.*, 1994, **50 (1),** 19.
278. Marjanovic, M., Pospisil, J. and Cikovic, N., *Kemija u Industriji*, 1994, **43,** 395.
279. Albertsson, A. C., Barenstedt, C., Karlsson, S. and Linberg, T., *Polymer*, 1995, **36 (16),** 3075.
280. Shah, P. B., Bandopadhyay, S. and Bellare, J. R., *Polym. Degrad. Stab.*, 1995, **47,** 165.
281. Sykes, M. L., Yen Hsuch-Wen, West, I. F., Gauldie, R. W. and Helsley, C. E., *J. Environ. Polym. Degrad.*, 1994, **2 (3),** 201.
282. Fanta, G. F. and Doane, W. M., in *Modified Starches: Properties and Uses*, Chap. 10, ed. O. B. Wurzburg. CRC Press, Boca Raton, FL, 1986.
283. Patil, D. R. and Fanta, G. F., *J. Appl. Polym. Sci.*, 1993, **47,** 1765.
284. Henderson, A. M. and Rudin, A., *J. Appl. Polym. Sci.*, 1982, **27,** 4115.
285. Dennenberg, R. J., Bothast, R. J. and Abbott, T. P., *J. Appl. Polym. Sci.*, 1978, **22,** 459.
286. Swift, G., *Polym. Mater. Sci. Eng.*, 1990, **63,** 846.
287. Fanta, G. F., Trimell, D. and Salch, J. H., *J. Appl. Polym. Sci.*, 1993, **49,** 1679.
288. Hocking, P. J., *J. Macromol. Sci. Rev. Macromol. Chem. Phys.*, 1992, **C32,** 35.
289. Bailey, W. J., *Prog. Pac. Polym. Sci. Proc. Pac. Polym. Conf.*, 1989, **1,** 105.
290. Bailey, W. J. and Kuruganti, V. K., *Polym. Mater. Sci. Eng.*, 1990, **62,** 971.

291. Feterpeker, S. A. and Potnis, S. P., *Angrew. Makromol. Chem.*, 1981, **93,** 111.
292. Lay, G., Rehm, J. and Stepto, R. F., US Patent No. 5,095,054, 1992.
293. Jane, J. L., Schwabacher, A. W., Ramrattan, S. N. and Moore, J. A., US Patent No. 5,115,000, 1992.
294. Willett, J. L., US Patent No. 5,087,650, 1992.
295. Liu, N. C. and Baker, W. E., *Adv. Polym. Technol.*, 1992, **11,** 249.
296. Pholman, A. A., MS dissertation, University of California, Berkeley, 1974.
297. Maldas, D. and Kokta, B. V., *Polym. Eng. Sci.*, 1991, **31,** 1351.
298. Raj, R. G. and Kokta, B. V., *Polym. Eng. Sci.*, 1991, **31,** 1358.
299. Vaidya, U. R. and Bhattacharya, M., *J. Appl. Polym. Sci.*, 1994, **52,** 617.
300. William, D. F., *Mater. Sci. Technol.*, 1987, **3 (10),** 797.
301. William, D. F., *Definitions in Biomaterials*. Elsevier, Amsterdam, 1987, p. 49.
302. Nakamura, T., Hitomi, S., Shimamoto, T., Hyon, S. H., Watanabe, S. and Shimizu, Y., in *Biomaterials and Clinical Applications*, ed. A. Pizzoferrato, P. G. Machetti, A. Ravaglioli and A. J. C. Lee. Elsevier Science, Amsterdam, 1987, p. 759.
303. Nakamura, T., Shimizu, Y., Matsui, T., Okumura, N., Hyan, S. H. and Nishiya, K., *Proc. 4th Int. Conf. on Biomaterials*. Denkendorf, 1992, p. 153.
304. Rehm, K. E., *Proc. 4th Int. Conf. on Biomaterials*. Denkendorf, 1992, p. 163.
305. Niu, S., Kurumatani, H., Satoh, S., Kanda, K., Oka, T. and Watanabe, K., *ASAIO Trans.*, 1993, **39,** M750–753.
306. Parsson, H., Jawien, A., Norgren, L. and Thorne, J., *VASA*, 1992, **21,** p6.
307. Greisler, H. P., Tattersall, C. W., Klosak, J. J., Cabusao, E. A., Garfield, J. D. and Kim, D. U., *Surgery*, 1991, **110,** 645.
308. Matsuda, T., Miwa, H., Moghaddam, M. J. and Iida, F., *ASAIO Trans.*, 1993, **39,** M327–M331.
309. Matsuda, T., Moghaddam, M. J., Miwa, H., Sakurai, K. and Iida, F., *ASAIO Trans.*, 1992, **38,** M154–M157.
310. Artandi, C., *Chemtech*., 1981, 476.
311. Unitica Co. Ltd, *Handbook for Application of New Functional Fibers*, ed. K. Chosakai. Sankyo Shuppan, Tokyo, 1988, p. 217.
312. Kuroyanagi, Y., Kim, E. and Shioya, N., *J. Burn Care Rehabil.*, 1991, **12,** 106.
313. Kuroyanagi, Y., Kim, E., Kenmochi, M., Ui, K., Kageyama, H., Nakamura, M., Ikeda, A. and Shioya, N., *J. Appl. Biomater.*, 1992, **3,** 153.
314. Koide, M., Osaki, K., Konishi, J., Oyamada, K., Katamura, T. and Takahasi, A., *Biomed. Mater. Res.*, 1993, **27,** 79.
315. Annas, I. V. and Burke, J. F., *J. Biomed. Mater. Res.*, 1980, **14,** 65.
316. Doillon, C. J., Whyne, C. F., Brandwein, S. and Silver, F. H., *J. Biomed. Mater. Res.*, 1986, **20,** 1219.
317. Boyce, S. T., Christian, D. J. and Hansbrough, J. F., *J. Biomed. Mater. Res.*, 1988, **22,** 939.
318. Yoshizato, K., Makino, A. and Nagayoshi, K., *Biomed. Res.*, 1988, **9,** 33.
319. Yasutomi, Y., Nakakita, N., Shioya, N. and Kuroyanagi, Y., *Burn Injuries*, 1993, **19,** 102.
320. Gebelein, C. G. and Carraher, C. E., *Polymeric Materials in Medication*. Plenum Press, New York, 1985.
321. Ensuminger, W. D. and Selam, J. L., *Infusion Systems in Medicine*. Futura, New York, 1987.
322. Duncan, R. and Kopecek, J., *Adv. Polym. Sci.*, 1984, **57,** 51.
323. Andreopoulos, A. G., *Clinical Mater.* , 1994, **15 (2),** 89.
324. Dubois, P., *Plastics in Agriculture*. Applied Science Publishers, London, 1978.
325. Jacopsohn, R. and Alon, H., *Weed Sci.*, 1980, **28,** 312.
326. Brighton, C. A., *5th Int. Coll. Comite Int. Plast*. 1972, p. 203.
327. Langren, B., *5th Int. Coll. Comite Int. Plast*. 1972, p. 233.
328. Maquin, M., *Plasticulture*, 1973, **18,** 38.
329. Buttrey, D., *Plast. Polym.*, 1969, **37 (127),** 65.
330. Buttrey, D., *Mod. Plast. Int.*, 1972, **2 (2),** 9.
331. Otey, F. H., *Polym. Plast. Technol. Eng.*, 1976, **7,** 221.
332. Westhoff, R. P., Otey, R. H., Mehltretter, C. L. and Russell, C. R., *Ind. Eng. Chem. Prod. Res. Dev.*, 1974, **13 (2),** 123.

333. Seymour, R. B., *Polym. News*, 1980, **6,** 101.
334. Kydonieus, A. F., (ed.), *Controlled Release Technologies: Methods, Theory and Applications*, Vols. I and II. CRC Press, Boca Raton, FL, 1980.
335. Paul, D. R., *Controlled Release Polymeric Formulations*, Amercian Chemical Society Symposium Series No. 33. ACS, Washington, DC, 1976, p. 2.
336. McCormic, C. L., Anderson, K. W. and Hutchinson, B. H., *J. Macromol. Sci. Rev. Mac. Chem. Phys.* , 1982, **22 (1),** 57.
337. Scher, H. B. (ed.), *Controlled Release Pesticides*, American Chemical Society Symposium Series No.53. ACS, Washington, DC, 1977.
338. Cardarellie, N. F., *Controlled Release Pesticides Formulations*. CRC Press, Boca Raton, FL, 1976.
339. Shasha, B. S., Trimnell, D. and Otey, F. H., *J. Polym. Sci., Polym. Chem. Ed.*, 1981, **19,** 1981.
340. Trimnell, D., Shasha, B. S., Wing, R. W. and Otey, F. H., *J. Appl. Polym. Sci.*, 1982, **27,** 3919.
341. Riley, R. T., *J. Agri. Food Chem.*, 1983, **31,** 202.
342. Shasha, B. S., Trimnell, D. and Otey, F. H., *J. Agric. Food Chem.*, 1981, **29,** 1099.
343. Foley, M. E. and Wax, L. M., *Weed Sci.*, 1980, **28,** 626.
344. Stout, E. L. and Doane, W. M., *J. Appl. Polym. Sci.*, 1979, **24 (1),** 153.
345. Scheiber, M. M. and White, M. D., *Weed Sci.*, 1980, **28,** 685.
346. McCormick, C. L. and Lichatowhich, D. K., *J. Polym. Sci.*, 1979, **17,** 479.
347. McCormik, C. L. *et al*., in *Controlled Release of Pesticides and Pharmaceuticals*, ed. D. H. Lewis. Plenum Press, New York, 1981, p. 147.
348. Kemp, M. V. and Wrightman, J. P., *Va. J. Sci.*, 1981, **32,** 34.
349. Baumley, J. J. and IInicki, R. D., *Proc. Annual Meeting, Northeast Weed Science Society*, Beltsville, MD, 1981, p. 52.
350. *Chemical Economics Handbook*. SRI International, Stanford, CA, 1981, p. 8001.
351. Hauck, R. D., in *Organic Chemicals in the Soil Environment*, 2nd edn, ed. A. I. Goring and J. W. Hamaker. Marcel Dekker, New York, 1972.
352. Hauck, R. D. and Koshino, M., *Fertilizer Technology and Use*, 2nd edn. Soil Science of America, Madison, 1971.
353. Sugimoto, K. J., *Ferment. Assoc. Japan*, 1990, **36,** 98.
354. Process for Producing Pullulan Fibers, European Patent, 1,496,017, 1975.
355. Nutricol Konja Flour, Marine Colloids Application Bulletin # k-2, 1989.
356. *Environment Today*, 1990, **1**(3), 3.