





Cite this: *Green Chem.*, 2022, **24**, 1762

Progresses and future prospects in biodegradation of marine biopolymers and emerging biopolymer-based materials for sustainable marine ecosystems†

François Samalens,^{‡a,b} Martin Thomas,^{‡a,b} Marion Claverie,^{a,b} Natalia Castejon,^{§a,b} Yi Zhang,^{a,b} Thierry Pigot,^a Sylvie Blanc ^a and Susana C. M. Fernandes ^{*a,b}

With approximately 250 000 marine species, the ocean is a vast reservoir of biodiversity and an abundant biological resource of natural polymers. The wide variety, renewable nature, tunable physicochemical and structural behavior and appealing biological properties make these marine biopolymers particularly attractive to the scientific community and numerous industrial sectors. As raw materials, they offer novel opportunities for the development of bio-based materials in response to recent demands for biodegradable plastic materials to lower plastic pollution in marine ecosystems. The biodegradation of marine biopolymers and biopolymer-based materials depends on marine environmental conditions such as temperature, pH and in particular microbial population. Marine microorganisms producing biopolymer-degrading enzymes (*i.e.*, hydrolases, lyases, oxidoreductases) are well studied, nonetheless the biodegradation processes of marine biopolymers-based materials in the marine/aquatic environment need further investigation. This review describes various biodegradation parameters and mechanisms of the degradation of marine biopolymers in the marine environment. It also puts emphasis on the marine microorganisms and the corresponding enzymes that catalyze the degradation of different marine biopolymers. Finally, it focuses on the few studies on biodegradation of emerging bio-based materials in aquatic ecosystems.

Received 20th November 2021,
Accepted 2nd February 2022

DOI: 10.1039/d1gc04327g

rsc.li/greenchem

1 Introduction

During the evolution over millions of years, marine organisms evolved extraordinary physical and chemical characteristics, which include the ability to biosynthesize and biodegrade natural polymers (also known as biopolymers) to support their survival.¹ Marine organisms namely algae, plants, animals and microorganisms can provide a large amount of marine biopolymers, which include proteins (*e.g.*, collagen) and polysaccharides (*e.g.*, chitin, chitosan, cellulose, alginate, *etc.*) for which the annual production represents 10¹² to 10¹⁴ tons.² On the other hand, these natural polymers can be converted to corresponding monomers by enzymes present in microbes, bacteria and fungi.³ Therefore, they have great potential to overcome the biodegradability issues related to the synthetic polymers dumped into the marine environment intentionally

or unintentionally. Natural polymers are attracting the interest of academics and industrials as biodegradable substitutes in plastics, materials and products where non-biodegradable and fossil-based polymers are currently used.⁴

Synthetic polymers are derived from petroleum-based sources and are mainly used for the manufacture of plastic products.⁵ Multiple industries including packaging and food packaging, building and construction, textiles, biomedical, electrical and electronics are producing millions of tons of plastic materials and disposable plastic products every year.⁶ Plastic production has increased exponentially, from 2.3 million tons in 1950 to 448 million tons by 2015; and it is expected to double by 2050. As consequence, an enormous amount of plastic trash (~57 million tons annually) is found and detected in several places in the Planet, including in the ocean.

Every year, at least 8 million tons of plastic end up in the ocean. These plastic products are originated from land-based plastic such as urban runoff, beach visitors, packaging, building and construction, textile sectors, and inadequate waste management, and from ocean-based plastic namely fishing industry, nautical activities and aquaculture.⁷ A major proportion of the plastics that has been found in the Great Pacific Garbage Patch (GPGP) is from fishing nets, ropes and lines sources.⁸

The rapidly increasing production of synthetic plastic products overwhelms the ability to deal with their recycling and

^aUniversité de Pau et des Pays de l'Adour, E2S UPPA, IPREM, CNRS, 64000 Pau, France. E-mail: susana.fernandes@univ-pau.fr

^bMANTA – Marine Materials Research Group, Université de Pau et des Pays de l'Adour, E2S UPPA, 64600 Anglet, France

†Electronic supplementary information (ESI) available. See DOI: 10.1039/d1gc04327g

‡These authors contributed equally to this work.

§Present address: Department for Food Chemistry and Toxicology, University of Vienna, Vienna 1090, Austria.

reuse, and in particular environmental (bio)degradation. For instance, most of the plastics made with synthetic polymers, such as olefin-based plastics, polyethylene (PE), polypropylene (PP), vinyl-based plastics such as polyvinyl chloride (PVC) and aromatic plastics (like polystyrene PS), and polyethylene terephthalate (PET) are resistant to environmental (bio)degradation.⁹ Because of their high molecular weight and unnatural structure, the biodegradation of synthetic polymers by microorganisms takes extremely long time (hundreds of years),¹⁰ and consequently, persist for long time in the environment.

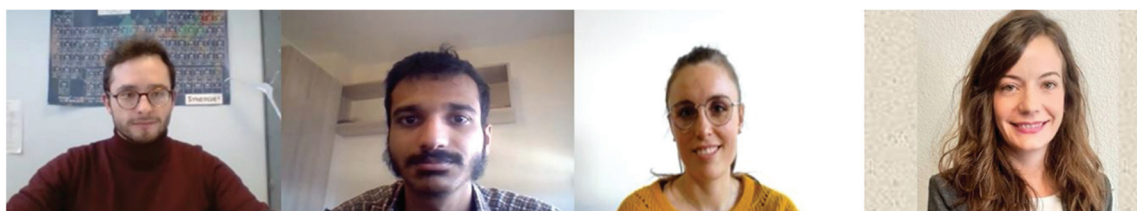
Also, under the effect of environmental factors like temperature, ultraviolet (UV) radiation, pH, salinity, currents and wind, these plastics are fractured into microplastics (particles < 5 mm) and nanoplastics (particles < 100 nm) causing a negative impact on marine wildlife and human health.^{11–13} Often marine organisms are entangled or ingest these plastic particles causing severe injuries and deaths, and also impacting seafood safety and quality and human health.^{11,12,14} Accordingly, plastic pollution has become one of the most pressing environmental and socioeconomic issues.

In this context, lower the devastating impact of the emerging pollutants in the marine environment is a major chal-

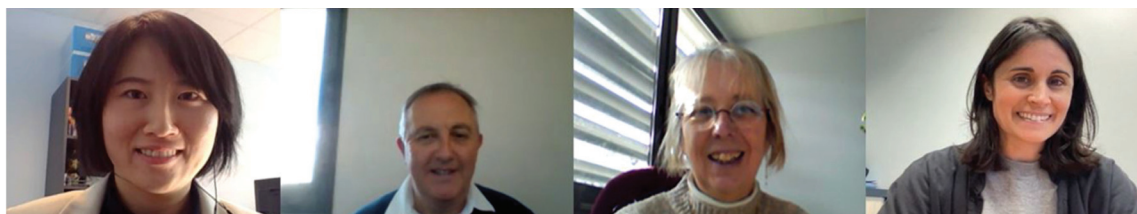
lenge in the 21st century. One possible solution, which has been investigated in the last decades, is the use of natural polymers, in particular derived from marine sources as an alternative to synthetic polymers for the manufacture of biopolymer-based materials and products.^{15–19} Table 1 lists the most investigated and used marine biopolymers for the production of biopolymer-based materials, their products and commercial brands as well as their field of application.

There is very little data available concerning the production of biopolymer-based materials. For instance, recently, Mintel Companies have estimated that 12 000 hyaluronic acid-based products were launched in 2019.⁴⁸ The statistics from the European Bioplastics Organization (<http://www.european-bioplastics.org/market>) show that bioplastics, which refer to plastics made from renewable biomass materials⁴⁹ represent less than 1% of the plastic produced annually. Nonetheless, growth is expected to increase greatly in the coming years, with projections showing the global production capacities in 2026 of approximately 7.59 million tons.

As it is a growing market, it is primordial to investigate the life cycle of the new biopolymer-based materials from their



François Samalens, Martin Thomas, Marion Claverie, Natalia Castejon



Yi Zhang, Thierry Pigot, Sylvie Blanc, Susana C. M. Fernandes

F. Samalens, PhD candidate, is working on polymer chemistry and cosmetics with the company Les Laboratoires de Biarritz and the research group MANTA.

M. Thomas, PhD candidate, is currently working in the photostability and impact of natural UV-absorbing molecules and their conjugates in aquatic environments.

Dr Claverie, has interests in enzyme characterization and engineering; enzymatic functionalization of biopolymers through a biomimetic approach for environmental and health applications.

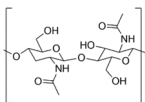
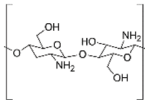
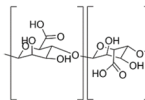
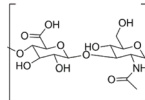
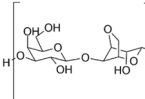
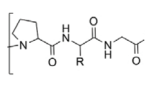
Dr Castejon, Marie Curie REWIRE Fellow, is working in novel cascading approaches based on green chemistry to valorize microalgal biomass by proving potential biological activities.

Dr Zhang, Assistant Professor, has research interests in enzymology, biocatalysis, marine proteins and bioinformatics.

Prof. Pigot, Full Professor, has interests in water quality and the reactivity of organic compounds in aquatic environments and photochemical processes of oxidation.

Dr Blanc, CNRS researcher, has interests in analysis of materials by UV visible and NIR spectral characterization taking into account reactivity and interface properties for environmental or health applications.

Dr. Fernandes, Associate Professor in Natural Polymers, is Chair in valorization of marine bioresources using green chemistry approaches and in development of marine bio-inspired materials. She is the PI of the research group MANTA.

Marine biopolymers and their structures		Origin	Products and brands	Fields of application	Ref.
Polysaccharides	Chitin 	Arthropods (shrimp, lobster, crab and insects) and mollusks	Anti-cholesterol agent, food preservative and food additive body creams, lotions, emulsifying agent, gelling agent, color stabilizer, thickener and stabilizer for sauces. Ultimate Miracle Worker Eye® Cream, Kristin Ess®, BST-Gel®, ChitoFlex® PRO, Protasan™, Reaxon® Shellworks	Water treatment and purification, food industry, packaging, agriculture, pulp and paper industry, cosmetics, tissue engineering and drug delivery	19–28
	Chitosan 				
	Alginate 	Brown seaweed, bacteria	Paper adhesion agent of tablets inject able fillers, antacids. Gaviscon Double Action® Peptac® Algycon® Maalox®	Pharmaceutical, drug delivery, wound healing, tissue engineering, food industry, textile, pharmacy, facial plastic surgery	29–33
	Hyaluronic acid 	Fishes	Films, and other wound dressings, dermal filler, lubricant. Contipro® gels Ordinary®	Medical and biological application, ophthalmology, tissue engineering, dermatology, cosmetics and treatment for osteoarthritis	34–38
	Agar 	Marine red algae	Texture improvement stabilizer, stabilizer for yoghurt, cheeses and candy. OBC Skin® Florence By Mills®	Food, biochemicals culture media for microbiology, electrophoresis, chromatography	39–43
Protein	Collagen 	Fishes, marine sponges and jellyfish	Skin and eye creams, Moisturizer drug supplements. Elems Pro-Collagen Marine® Collagen cosmetics®	Biomedical and cosmetic sectors, tissue engineering and cosmetics.	44–47

With the trend of using marine biopolymer-based materials to replace synthetic plastic in daily life, this review aims: (i) to describe the biodegradation parameters in marine ecosystems; (ii) to do a biodiversity map of relevant microorganisms and corresponding enzymes identified in marine ecosystems; (iii) to report the biodegradation process of biopolymers in marine ecosystems; (iv) to depict the biodegradation mechanisms of marine biopolymers (*e.g.* chitin, chitosan, alginate, collagen and hyaluronic acid) by hydrolases, lyases and oxidoreductases; and (v) to give an overview of the current researches and the limitations about the (bio)degradation of marine biopolymers and their bio-based materials in the marine environment.

2 (Bio)degradability of (bio)polymers in marine ecosystems: general aspects

Polymer degradation is extremely dependent on the surrounding environment. Thus, abiotic factors like water salinity, depth, temperature, flow, and biotic factors like microorganisms biodiversity composition are environmental factors to

take into consideration.⁵⁶ Consequently, polymer (bio)degradation is dependent of the aquatic ecosystems generally classified as: freshwater, estuaries, and marine ecosystems.

Freshwater ecosystems have a small amount of salt dissolved in water. They are often divided into two categories: the ones in which the water is stationary like lakes and ponds, and the ones in which there is a flow of water like rivers. In stationary water ecosystems, the conditions are heterogeneous affecting their populations. For instance, deep oligotrophic lakes have clear and cold water with a low amount of nutrients. Consequently, the microorganisms tend to be less productive. On the other hand, eutrophic lakes are shallow, nutrient rich and warm. Therefore, the productivity is different and it affects the potential biodegradability of organic materials. The microbial diversity of freshwater ecosystems is dominated by cyanobacteria and microalgae.⁵⁷

Estuaries are partially enclosed areas located at the junction between freshwater ecosystems and marine ecosystems (ocean). Unlike in marine or freshwater ecosystems, the amount of salt dissolved is not constant and it changes with the flow of water. Because of these particular conditions, the number of microorganism species present in estuaries is lower than that in marine or freshwater ecosystems.⁵⁸

Marine ecosystems are characterized by the presence of a high salt content (higher than in freshwater ecosystems and estuaries). These ecosystems are classified according to the distance to the coast and to the water depth. There are two main domains: Benthic and Pelagic (Fig. 1). The Pelagic Domain or open ocean waters correspond to the zones that are not in contact with the seabed (Fig. 1). The organisms living there are not attached to the seabed of the ocean (or sea). In these zones, the majority of organisms that carry out photosynthesis are microscopic: microalgae or bacteria. The principal factor that influences the type of microbial community is the amount of dissolved inorganic matter (nutrients).

Benthic Domain, zones located at the interface with the sedimentary deposits of the seabed (Fig. 1), contains a great quantity of nutrients and, consequently the biggest activity of microorganisms. Sponges, crustaceans or seaweed are commonly found in these areas. The population highly depends

on temperature and substrate present at the seabed (e.g., sand, mud). For instance, coral reefs or mangrove swamps are only found in warm Benthic Domains.⁵⁹ Most of biodegradation is achieved at the interface of water and sediments because it contains a large quantity of materials that are able to support the growth of microorganisms. According to the water depth, Benthic Domain is often divided as follows: the littoral zone; the sublittoral zone (between 0 to 200 m) where it has been found that some microorganisms like *Streptomyces* sp. *D1* release amylase;⁶⁰ the bathyal zone (between 200 to 2000 m) where microorganisms like *Thalassomonas* sp. *JAMB-A33* release agarase;⁶¹ the abyssal zone (between 2000 to 6000 m) where the quantity of nutrients are low due to the absence of light;⁶² and the hadal zone (below 6000 m).

Depending on the marine ecosystem, the activity of microorganisms' changes and therefore, the biodegradation kinetics. A study compared the activity of hydrolytic enzymes (e.g., chitinase, lipase and β -glucosidase) in different marine environments, and it found that the enzymatic activity was higher in shallow water sediments than that in deep-sea sediments, while the activity exponentially decreased with the sediment depth.⁶³

Since our focus is on marine ecosystems, the biodegradation of polymeric matter will be discussed with environmental conditions like temperature and pH which are major factors influencing the microorganism's proliferation and activity in marine ecosystem.

2.2 Biodegradation factors in marine ecosystem

2.2.1 Effects of environmental parameters. Environmental conditions at specific pH and temperature affect the bacterial population and, consequently the properties of the enzymes.⁶⁴ It has been demonstrated that slight changes in pH values (less than 1) can lead to a modification of the bacterial community. Moreover, some enzymes derived from marine bacteria living in extreme conditions (extremophiles) show the highest activity at extreme temperature and pH. For instance, an amylase obtained from a bacteria in Antarctica had maximum activity at around 10 °C.⁶⁵

On the other hand, some (bio)polymers-degrading enzymes show optimal catalysis at non-extreme conditions. For example, the biodegradation of bio-sourced polymers like poly (3-hydroxybutyrate) (P(3HB)), belonging to the family of polyhydroxyalkanoates (PHA), by bacteria *Pseudomonas stutzeri* *YM1414* present in lake water was investigated.⁶⁶ It was found that at pH of 9.5 and temperature of 55 °C, the depolymerases secreted by the bacteria had the highest activity for the degradation. Various alginate lyases from molluscs also showed the highest activity at pH values from 5.6 to 8.5 and temperature ranges between 35 to 53 °C.⁶⁷

2.2.2 Effects of (bio)polymer properties. Biodegradation process is also dependent on the physico-chemical properties of (bio)polymers. Surface properties (e.g., surface area, roughness), physico-chemical properties (e.g., chemical structure, molecular weight and distribution, crystallinity, crystal structure, amorphous nature), thermal properties (e.g., glass tran-

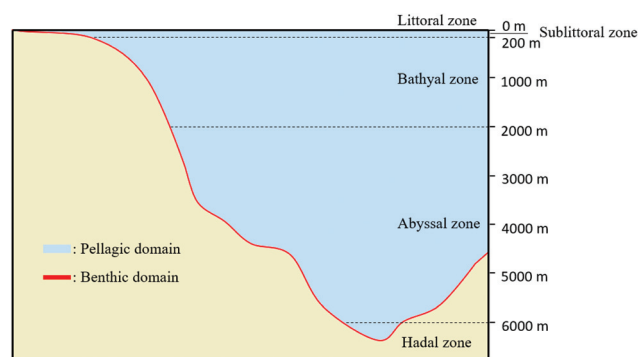


Fig. 1 Representation of the two main domains of marine ecosystems: Benthic Domain and Pelagic Domain (inspired by ref. 59).

sition and melting temperatures), mechanical properties (e.g., modulus of elasticity), hydrophilicity, hydrophobicity, shape and porosity of the (bio)polymers are all the factors that can influence biodegradation processes.⁶⁸

All these properties have been studied for the biodegradation of synthetic polymers, however, only few studies have been done regarding natural polymers. For instance, the biodegradation of cellulose was greater when it had a low degree of crystallinity.⁶⁹ Biodegradation of natural polymer chitin by enzymes (like lysozyme or lipase) is variable according to the structure of chitin whiskers (are nanocrystals of chitin obtained after removing the amorphous domains of the biopolymer).⁷⁰ When the deacetylation degree of chitin increases, the biodegradation rate increases. Liu *et al.* described that the presence of amino groups allowed a better combination between the enzymes and the chitin whiskers⁷⁰ that helps with the enzymatic biodegradation. Besides, the shape of (bio)polymers in marine ecosystems affects its biodegradation. It was demonstrated that PHA films were degraded faster than PHA pellets due to their larger surface area. The larger polymer/water interface improves the adhesion of the microorganisms to the surface of the polymer.⁷¹

2.3 Biodiversity map of relevant microorganisms and enzymes in marine ecosystem

A wide variety of microorganisms like anaerobes, aerobes and photosynthetic bacteria as well as fungi present in marine ecosystems are able to degrade synthetic and natural polymers by releasing molecules and/or enzymes. These microorganisms are diverse, for example, it is possible to find hundreds of millions of bacteria per gram of marine sediment only.⁷² The degradation of synthetic polymers by microorganisms has recently been

reviewed.^{54,73} Recently, Sheth *et al.* reviewed 50 different species of marine bacteria, fungi and enzymes that have been isolated to degrade synthetic polymers like Nylon, PET (polyethylene terephthalate), PU (polyurethane), PE (polyethylene), PVC (polyvinyl chloride).⁵⁴ Nonetheless, the research on polymer-degrading organisms is still scarce. Herein, for the first time, a biodiversity map regarding the location of the different extracted marine biopolymers-degrading enzymes is presented in Fig. 2 and an updated list in Table 2 (and ESI Table S1†). The enzymes that are listed belong to the Carbohydrate-Active enZYmes database (CAZy, <http://www.cazy.org>) which list and classify the enzymes that catalyze the breakdown of polysaccharides (marine and non-marine). The present list of biopolymer-degrading organisms and enzymes was obtained by reviewing the current literature using different database like Scopus and Google Scholar with the keywords: 'enzyme', 'degradation', 'degrading', 'marine', 'biopolymer', 'natural polymer', 'organism', 'polysaccharide', 'agar', 'starch', 'chitin', 'chitosan', 'cellulose', 'alginate', 'pullulan', 'xylan', 'hyaluronic acid', 'collagen'. As displayed in Fig. 2, the identified microorganisms and their corresponding enzymes are mostly from Japan, Pacific Ocean, Europe and Antarctica.^{60,61,65,74–104} The identified microorganisms were isolated from marine organisms (in orange pastel color), water (Freshwater or Seawater, in blue color) or sediments (in green pastel color) from the Benthic Domain, and most of the responsible enzymes are hydrolases from bacteria (Fig. 2 and Table 2).

As showed in Fig. 2 and listed in Table 2, there are diverse marine microorganisms from different marine ecosystems producing enzymes that are able to degrade natural polymers in diverse conditions. For instance, the marine organisms known as extremophiles, organisms that live in extreme conditions of temperature and pH, present different properties from those

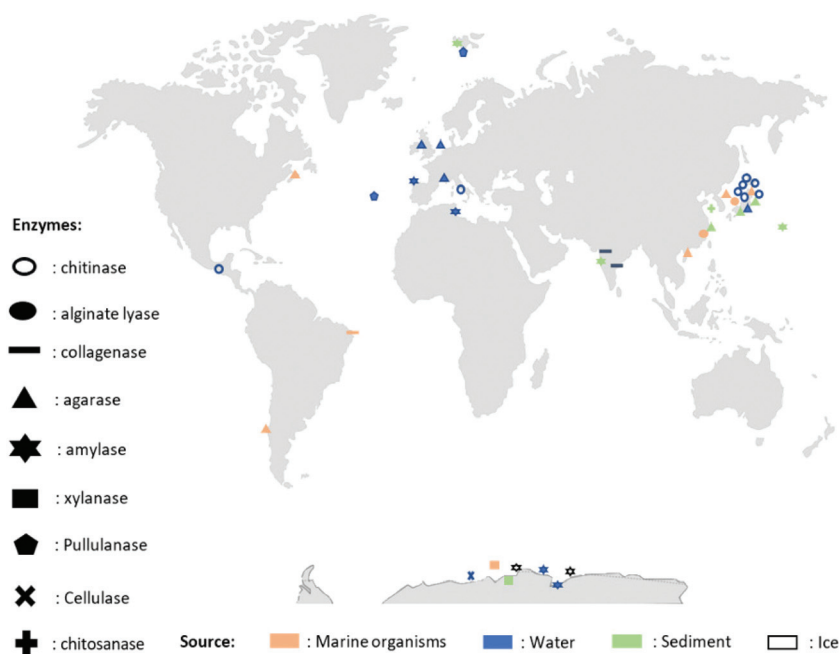


Fig. 2 Map representing different enzymes degrading biopolymers, their extraction location and their source.

Table 2 List of microorganisms and enzymes degrading polysaccharides and proteins: agar, starch, chitin, chitosan, cellulose, alginate, pullulan, xylan, hyaluronic acid, collagen in marine environments

Polysaccharides		Proteins	
Microorganisms	Associated enzymes	Microorganisms	Associated enzymes
<i>Vibrio</i> sp. JT0107 <i>Cytophage</i> sp. <i>Alteromonas agarlyticus</i> GJ1B <i>Vibrio</i> sp. PO-303 <i>Agarivorans</i> sp. HZ105 <i>Alteromonas</i> sp. SY37-12 <i>Pseudoalteromonas antarctica</i> N-1 <i>Pseudomonas atlantica</i> <i>Vibrio</i> sp. AP-2 <i>Agarivorans albus</i> YKW-34 <i>Cytophage flevensis</i>	Agarase	<i>C. leiarchus</i> <i>Pseudomonas</i> <i>Pseudomonas marinoglutinosa</i>	Collagenase
<i>Streptomyces</i> sp. D1 <i>Aureobasidium pullulans</i> N13d <i>Pseudoalteromonas haloplanktis</i> <i>Haloferax mediterranei</i> <i>Pseudoalteromonas</i> sp. M175 <i>Glaciozyma antarctica</i> PI12 <i>Alteromonas</i> sp. TAC 240B	Amylase		
<i>Clonostachys rosea</i> <i>Stenotrophomonas maltophilia</i> <i>Vibrio fluvialis</i>	Chitinase		
<i>Vibrio parahaemolyticus</i> <i>Vibriogalginolyticus</i> <i>Alteromonas hydrophila</i> <i>Vibrio mimicus</i> <i>Listonella anguillarum</i>	Chitinase/chitobias		
<i>Bacillus</i> sp. Q1098 <i>Pseudoalteromonas haloplanktis</i>	Chitinase Cellulase		
<i>Cobetia</i> sp. NAP1 <i>Microbulbifer</i> sp. ALW1	Alginate lyase		
<i>Shewanella arctica</i> <i>Fervidobacterium pennavorans</i>	Pullulanase		
<i>Cladosporium</i> sp. <i>Fervidobacterium frigidarium</i> sp.	Xylanase		
<i>Vibrio</i> sp. FC509	Hyaluronidase		

Biopolymers	
	: Agar
	: Starch
	: Chitin
	: Chitosan
	: Cellulose
	: Alginate
	: Collagen
	: Xylan
	: Pullulan
	: Hyaluronic acid

living in “normal conditions”. As example, extremophiles are: (i) psychrophiles living in environments of low temperature (between -2 and 20 °C); (ii) thermophiles living in high temperature mediums (between 55 and 113 °C); (iii) acidophiles living in acidic mediums ($\text{pH} < 4$); and (iv) alkaliphiles living in basic mediums ($\text{pH} > 9$).¹⁰⁵

The enzymes that can break down chemical bonds between monomers of natural polymers are mainly hydrolases and lyases. Chitinases (biosynthesize by *Vibrio fluvialis* and *Vibrio parahaemolyticus*⁷⁴), alginate lyases (by *Microbulbifer* sp. ALW and *Cobetia* sp. NAP1^{75,76}) or agarases (by *Vibrio* sp. JT0107 and by *Alteromonas agarlyticus* GJ1B^{84,86}) have been reported. Regarding extremophilic organisms, *Alteromonas* sp. TAC 240B, a psychrophile found in Antarctica, produced amylase, an enzyme degrading starch;⁷⁷ *Flavobacterium frigidarium* sp. was found in Antarctica producing xylanase, an enzyme degrading xylan.⁷⁸

2.4 Biodegradation process of (bio)polymers in marine ecosystems

The biodegradation process *via* the action of microbial enzymes occurs through a sequence of steps, *i.e.* biodeteriora-

tion (initial change of physical and chemical properties of the polymer), bio-fragmentation (disintegration of the polymeric structure into smaller and simpler fragments *via* enzymes) and bioassimilation (ingestion of molecules by microorganisms),¹⁰⁶ as described in Fig. 3.

2.4.1 Biodeterioration. The deterioration process of the polymers in the marine environment is considered to be the same whether it is synthetic or natural. It is a combination of abiotic (caused by the environmental conditions) and biotic factors (caused by the action of living organisms) leading to the fragmentation of the polymer chains and a deterioration of the global shape of a material, resulting in the structure deterioration.^{107,108}

Microorganisms like bacteria, algae or fungi can deteriorate the polymers mechanically, chemically or enzymatically.^{10,109} The microorganisms deteriorate (bio)polymers in a physical way to increase the size of the pores in polysaccharides and proteins to weaken the material.^{110,111} Some microorganisms like chemolithotrophic or chemoorganotrophic bacteria can degrade polymers in a chemical way (*i.e.*, *Nitrobacter* spp.). They produce acids (*e.g.*, nitric acid, sulphuric acid) known to deteriorate organic matter. These two families of

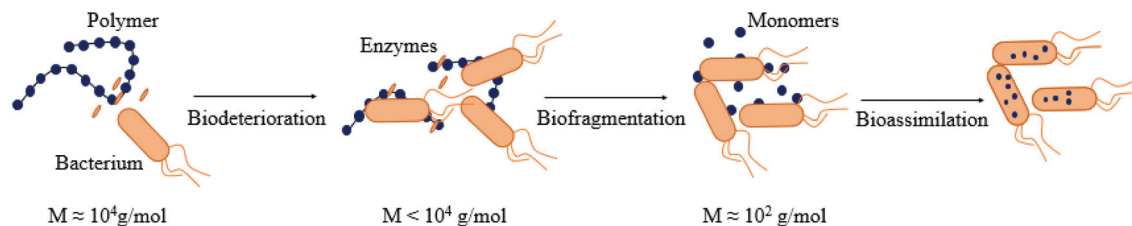


Fig. 3 Different steps of biodegradation of (bio)polymers by bacteria: biodeterioration, biofragmentation, bioassimilation. (inspired by ref. 54).

microorganisms are different in terms of the way that they obtain energy to produce acids. Chemolithotrophic bacteria use inorganic sources whereas chemoorganotrophic microorganism use organic matter.^{112–114} As the biodeterioration goes on, the water in environment cause swelling of the (bio)polymers, facilitating the degradation. Moreover, (bio)polymers can also undergo biodeterioration by enzymes produced by marine microorganisms. Even synthetic polymers like polyvinylchloride and polyamide that are known to be hardly degradable¹¹⁵ could be deteriorated by enzymes from marine organisms like esterases or proteases.¹¹⁶ For instance, Webb *et al.* showed that plasticized polyvinyl chloride was deteriorated by esterases.¹¹⁷ The degradation of polymers derived from natural sources like poly(L-lactide) in seawater was studied with enzymatic biodeterioration which was described as a bulk erosion of the polymers.¹¹⁸ There are two types of erosion when it comes to enzymatic biodeterioration: the surface erosion traduces a loss of matter but no change in the molecular weight; and the bulk erosion happens when the molecular weight of the polymer is reduced due to bond cleavage.¹¹⁹ However, the bulk erosion can belong to biofragmentation as it breaks chemical bonds.

2.4.2 Biofragmentation and enzymatic hydrolysis. Biofragmentation is the process that reduces the molecular weight of a (bio)polymer by cleaving bonds to produce oligomers and monomers. This step is necessary to allow bioassimilation in the microbial cells because full-size polymers are unable to cross cell membranes.

There are many different microorganisms taking action during this step. They secrete specific enzymes, and each one has specific function. There are two main reactions during biofragmentation: enzymatic hydrolysis and enzymatic oxidation. The hydrolases in this step are capable of converting the carbohydrates into sugars, lipids into long chain fatty acids, and proteins into amino acids.^{120,121} Taking polysaccharides as an example, enzymatic hydrolysis reaction of the glycosidic bond is catalyzed and usually requires a proton donor (HA), and a nucleophile or a base (B⁻). If the polymer is hardly hydrolyzed because of its crystallinity, oxidoreductases (mono-oxygenases, di-oxygenases, oxidases) can take part in the biofragmentation step. They are capable of oxidizing polymers to create alcohols.

2.4.3 Bioassimilation. After the biofragmentation step, the biodegradation of the material ends with the bioassimilation (Fig. 3). The monomers formed during the bioassimilation depends on the type of microorganism. The monomers can cross the cellular membrane of microorganisms giving the

cells sources of energy to grow (*e.g.*, carbon, oxygen...). The molecules entering the cells are oxidized and participate in the formation of adenosine triphosphate (ATP). The molecules can be assimilated by organisms using aerobic respiration or anaerobic respiration.¹⁰⁷

3 Biodegradation mechanisms of marine biopolymers

An increasing number of novel materials are made up from natural polymers synthesized by marine organisms. They have been used in various applications, including in medical or cosmetic products.^{122,123} Similar to the fate of synthetic polymers (*e.g.*, plastic), the novel marine biopolymer-based materials can also end up in the ocean, therefore, it is important to understand their biodegradation mechanisms and determine their impact on marine ecosystems.

3.1 Biodegradation mechanisms by hydrolases and lyases

As described previously in section 2.3, hydrolases and lyases are the two major enzyme classes (EC 3 and EC 4, respectively) that can catalyze the degradation of marine biopolymers. Herein, the biodegradation mechanisms of principal marine polymers including polysaccharides namely chitin and chitosan, alginate, hyaluronic acid, as well as a protein, collagen are summarized and compared.

3.1.1 Biodegradation of marine polysaccharides

3.1.1.1 Chitin and chitosan. Chitin is a linear long-chain polysaccharide composed of *N*-acetylglucosamine units linked via β -(1 \rightarrow 4) bonds.¹²⁴ Chitin is considered the most abundant biopolymer in the biosphere after cellulose.¹²⁵ Chitin is present in arthropods (shrimp, lobster, crab and insects) and mollusks (squid pen). Moreover, in the marine environment a variety of unicellular eukaryotic organisms synthesize chitin such as ciliates, cnidosporida, rhizopoda, diatoms, yeast and fungi.^{126–128} Its chemical structure is similar to cellulose but chitin contains predominantly acetamide groups ($-\text{NHCOCH}_3$) or residual amine groups ($-\text{NH}_2$) at the C-2 position. In nature, chitin exists as ordered crystalline microfibrils wrapped in protein and embedded in minerals like calcium carbonate and residual calcium phosphate. It is insoluble in water and in most organic solvents.¹²⁹ Chitin is present in three different polymorphic forms α -, β - and γ -chitin that have different packing and polarities of near chains in the successive sheets

of which they are composed. It occurs in different morphological forms in living organisms such as small microfibrils with diameter of 2.5–3 nm (in arthropod cuticles), large microfibrils 9 to 27 nm in diameter and up to 1 μm length (in fungi Vestimentifera worms) and nematic liquid crystal structure (in Pogonophora). Due to its abundance and unique physico-chemical and biological properties like biocompatibility, antimicrobial activity, and biodegradation, chitin is becoming one of the most important chemical raw materials for the fabrication of emergent sustainable polymer materials. In the last decades, important advances have been reported in: chitin extraction and solubilization,^{15,130,131} use of chitin as functional materials for environment depollution,^{132–134} chitin nanocrystals/nanofibers and polyols synthesis^{17,131,135–139} and characterization,¹⁸ as well as biomimetic materials design for tissue engineering.^{140,141}

Chitosan (β -1,4-linked glucosamine) is the main chitin derivative, and it has emerged as a relevant bio-based polymer for the fabrication of novel sustainable materials. Chitosan is obtained *via* the deacetylation of chitin with concentrated sodium hydroxide. It is considered that when the degree of deacetylation is higher than 40–60%, the biopolymer is called chitosan (corresponding to the fraction for which the polymer becomes soluble in acidic solutions). Chitosan has many applications such as water treatment and purification (removal of heavy metal ions and dyes),²⁰ in food industry as anti-cholesterol agent, food preservative and food additive,²¹ packaging,²² agriculture (seed and fertilizer coating),²³ pulp and paper industry (surface treatment, adhesive paper),^{19,24} cosmetics (body creams, lotions, *etc.*),²⁵ tissue engineering and drug delivery.²⁶

The biodegradation of chitin and chitosan as such has been found with function of endoenzymes and exoenzymes. There are two pathways of chitin biodegradation characterized: chitinolytic pathway and chitosan pathway (Fig. 4(a)).¹⁴²

In the chitinolytic pathway, the depolymerisation of chitin is realized by endochitinases (chitinase) and exochitinases (chitobiase and chitobiosidase). Endochitinases hydrolyze the chitin resulting in the formation of oligosaccharides of different chain lengths, however this *endo*-hydrolysis does not occur on small chains containing less than three acetyl glucosamine residues. Endochitinases cleave bonds randomly along the chitin strand and form loose ends. It is worth noting that the rate of hydrolysis is directly in proportional to the degree of polymerization of the chitin chains.¹⁴³ After the explosion of a number of chain ends, exochitinases catalysis allow the production of disaccharides, transforming chitin into chitobiose.¹⁴⁴ Subsequently, *N*-acetyl- β -D-glucosaminidase converts these disaccharides into *N* acetyl-glucosamine (GlcNAc). The monosaccharides products can be directly absorbed by cells. Finally, the glucosamine *N*-acetyl transferase catalyzes the conversion of *N*-acetylglucosamine to glucosamine (Fig. 4(b)).¹⁴⁵

In the chitosan pathway, there is a partial or total deacetylation step, where chitin deacetylase change chitin into chitosan. Chitosan is then hydrolyzed by chitosanase to oligomers of glucosamine ((GlcN)₂) (Fig. 4(c)). These oligomers are hydro-

lyzed by glucosaminidase, yielding to free glucosamine residues that can be used as direct substrates by cells.¹⁴⁶

The degree of deacetylation of chitin and chitosan have a great impact on the biodegradation. For instance, chitosan with low degree of deacetylation (<65% of acetylated units) was proven to be degraded by chitinases.¹⁴⁷ Chitinase are mainly synthesized by bacteria and fungi, which ingest chitin in their diet. The marine organisms that produce chitinase and chitobiase include *Vibrio fluvialis*, *Vibrio parahaemolyticus* or *Clonostachys rosea*^{74,80} (Fig. 2 and Table 2). Chitosanase hydrolyze the breakdown of chitosan.¹⁴⁸ This enzyme has been found in 25 types of fungi and 15 types of bacterial strains comprising marine bacteria such as *Bacillus* sp. and *Pseudoalteromonas* sp. SY39.^{149,150}

3.1.1.2 Alginate. Alginate is a polysaccharide formed by linear block copolymerization of (1 \rightarrow 4)-linked β -D-manuronic acid (M block) and α -L-guluronic acid (G block).¹⁵¹ The most important source of this natural polymer is brown seaweed, including *Phaeophyceae*, *Laminaria hyperborea*, *Laminaria digitata*, *Laminaria japonica*, *Ascophyllum nodosum* and *Macrocystis pyrifera*.¹⁵¹ Moreover, alginates are also synthesized by some bacteria such as *Pseudomonas aeruginosa* and *Azotobacter vinelandii*. Its intrinsic properties make alginate, one of the most promising biopolymers for a variety of applications in the pharmaceutical,³⁰ drug delivery,³¹ wound healing,³² tissue engineering³³ and food industry.¹⁵² Alginates are widely used due to their biodegradability, biocompatibility and low toxicity as well as their low extraction and purification costs. In addition, alginate can be processed into various forms such as matrices, hydrogels, particles and beads that make it very attractive.

Alginate is naturally degraded by enzymes, alginate lyases that can be produced by marine bacteria, such as *Cobetia* sp. *NAP1* and *Microbulbifer* sp. *ALW1*,^{75,76} (Fig. 2 and Table 2). These enzymes act on the glycosidic linkage of alginates, to produce polysaccharides of different carbon chain lengths (Fig. 5).¹⁵³

The alginate degrading enzyme complex consists of at least two different enzyme components: alginate lyase (eliminase) and *endo*-alginate hydrolase. These enzymes reduce the viscosity of alginate by scissoring the polymer chains resulting in the formation of saturated and unsaturated uronic acid with non-reducing groups.¹⁵⁴ The unsaturated mannuronic acid can be converted to 4-deoxy 5-keto uronic acid *via* tautomerism.

3.1.1.3 Hyaluronic acid. Hyaluronic acid (hyaluronan, HA) is a linear high molecular-weight polysaccharide composed of D-glucuronic acid and *N*-acetyl-D-glucosamine units linked by (1 \rightarrow 4) and (1 \rightarrow 3) bonds.³⁴ Each repeating disaccharide unit has one carboxylate group, four hydroxyl groups and an acetamido group. It belongs to the glycosamino glycans (GAGs) category of substances that include chondroitin sulphate, dermatan sulphate and heparin sulphate. Unlike other members in the GAG group, HA is devoid of sulphate group. It is highly hydrophobic with lubricating property. In terms of configuration, it exists as random coils that entangle at very high mole-

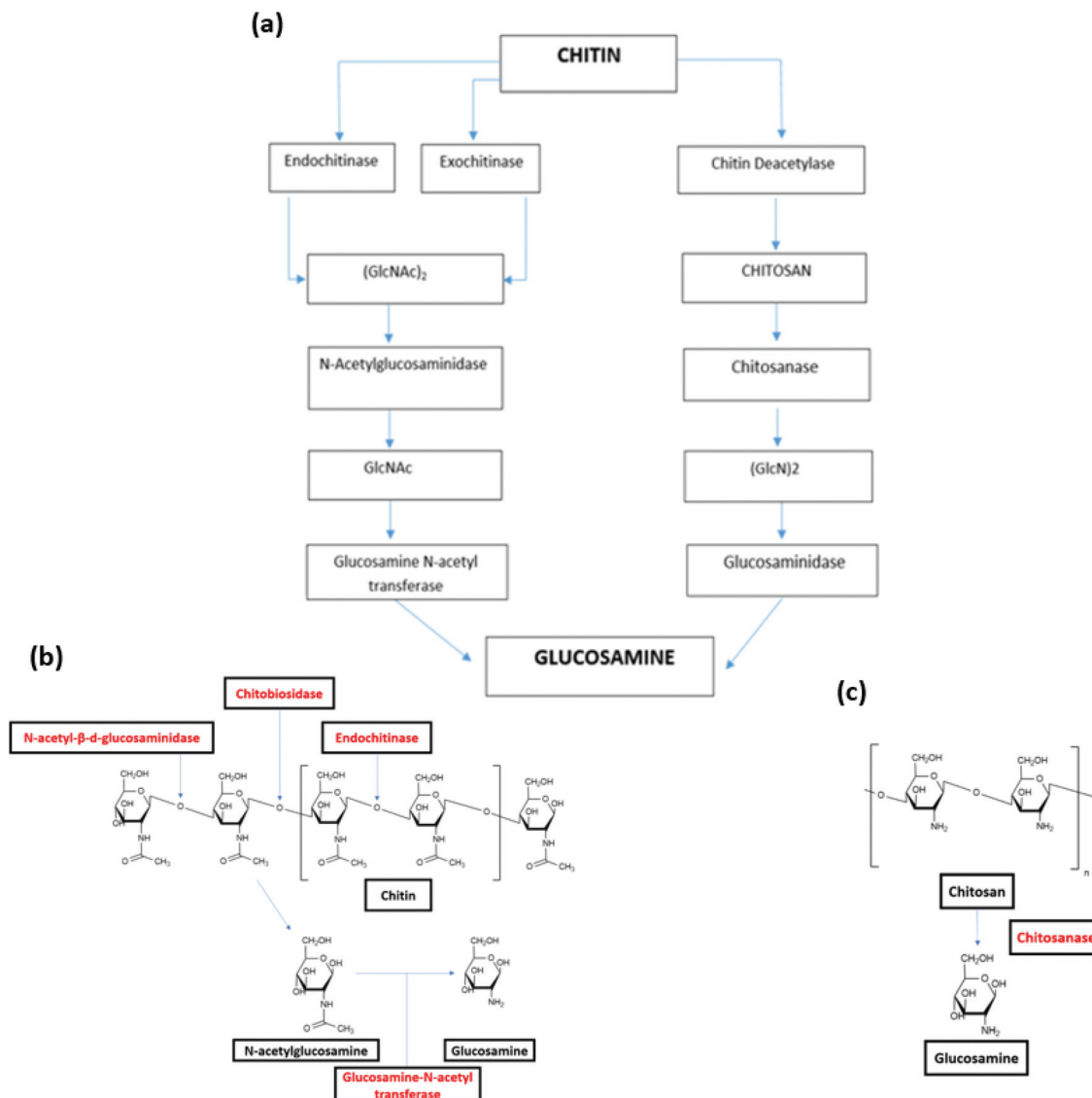


Fig. 4 (a) Pathways of chitin biodegradation in marine environments via the chitinolytic and chitosan pathway.¹⁴² (b) Degradation of chitin by endoenzymes and exoenzyme.¹⁴⁶ (c) Degradation of chitosan by chitosanase.¹⁴⁶ (a) Reprinted with permission from ref. 142, copyright American Chemical Society, 1998. (b) Reprinted with permission from ref. 146, copyright Springer Nature, 2015. (c) Reprinted with permission from ref. 146 copyright Springer Nature, 2015.

cular weight to form viscoelastic gels. HA is abundantly available in living organisms especially the soft connective tissues. In marine organisms, HA can be found in cartilages and vitreous humor of fish.¹⁵⁵ HA has important medical and biological applications due to its nonimmunogenic nature. Its biocompatibility and biodegradability make it useful in, ophthalmology,³⁵ tissue engineering,³⁶ dermatology,³⁷ cosmetics³⁸ and treatment for osteoarthritis.³⁴

Hyaluronic acid is degraded by enzymes such as hyaluronidase (hyaluronate lyase), β-D-glucuronidase and β-N-acetylhexosaminidase.^{156,157} The enzyme hyaluronidase cleaves the high molecular weight hyaluronic acid into smaller oligosaccharides. Subsequently, the β-D-glucuronidase and β-N-acetylhexosaminidase degrade the oligosaccharide fragments by

removing non reducing terminal sugars.¹⁵⁸ The degradation of hyaluronate lyase primarily produce unsaturated disaccharide and 2-Adgpaug (2-acetamido-2-deoxy 3-O-(β-D-glucosyl-4-enepyranosyluronic acid)-D-glucose) as the final products (Fig. 6).¹⁵⁹ In marine environment, the only bacteria identified that releases hyaluronidase is *Vibrio* sp. FC509.¹⁶⁰

3.1.1.4 Agar. Agar is a complex polysaccharide found in the cell walls of marine red algae. The structure of agar varies depending on the source, but it is commonly considered that this polysaccharide consists of mainly 3,6-anhydro-L-galactoses, D-galactoses and L-galactoses units linked by β(1,4) and α(1,3) linkages. The main chemical structure of agar is agarose and the main repeating moieties are 3-O-linked β-D-galactopyranose (G) and 3,6-anhydro-α-L-galactose (LA).¹⁶¹ Agar

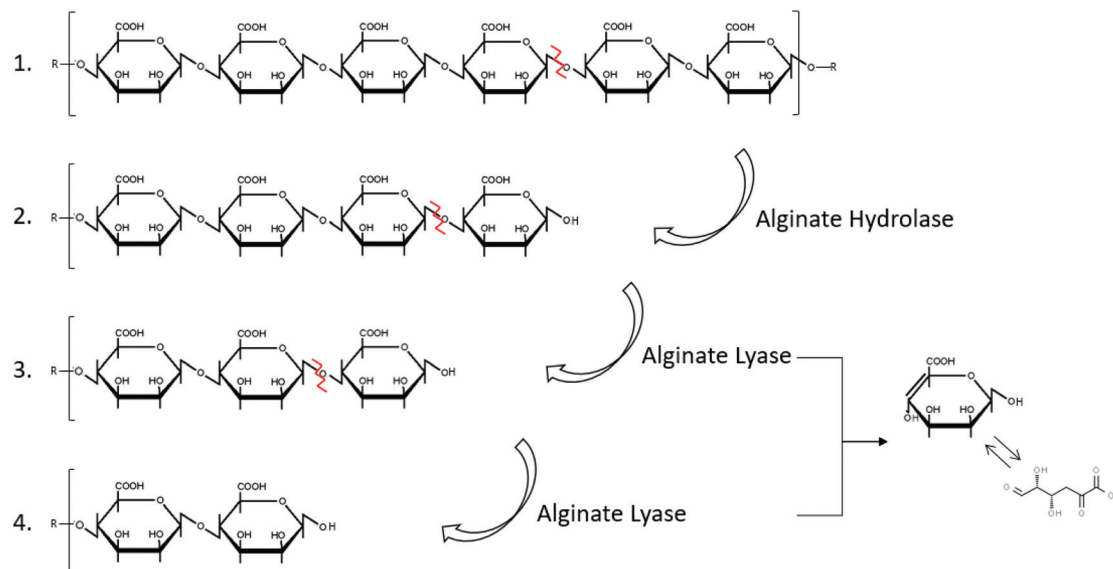


Fig. 5 Degradation of alginate by the enzymes hydrolase and lyase (inspired by ref. 154).

may also refers to moieties called porphyrobiose consisting of 3-O-linked β -D-galactopyranose (G) and α -L-galactose-6-sulfate (L6S). Due to the gelling properties, agar is used in various industries such as food (food additive⁴⁰) and biochemicals (culture media for microbiology⁴¹), and agarose gel electrophoresis⁴² and chromatography.⁴³

The complete mechanism of biodegradation of agar by marine organisms and enzymes has been reviewed by Chi *et al.*¹⁶² Herein, the focus of this review is on the degradation of agarose by marine enzymes. The biodegradation of agarose occurs in two pathways depending on if the structure is α -agarose or β -agarose. Agarose is initially cleaved by the enzymes α - or β -agarase at the positions α -(1,3) or β -(1,4), releasing agarotetraose or neoagarotetraose, respectively. Then, these products are broken down to agarobiose (also called neoagarobiose). Finally, agarobiose hydrolases cleave the β -(1,4) or the α -(1,3) bonds to form the monomeric units 3-O-linked β -D-galactopyranose (G) and 3,6-anhydro- α -L-galactose (LA), respectively. The porphyrobiose moieties are further biodegraded in a different pathway called the β -porphyran hydrolytic pathway involving other enzymes like β -porphyranase that hydrolyzes porphyran at the β -(1,4) linkage. There have been several agarose enzymes reported from marine bacteria in sea water, marine sediments or marine algae and mollusks. For instance, *Alteromonas agarlyticus* GJ1B in seawater,⁸⁶ *Agarivorans* sp. HZ105 from marine sediments⁸⁸ and *Alteromonas* sp. SY37-12 from red algae⁸⁹ were reported to express agarase. The Fig. 7 represents one of the possible agarose degradation pathways by a freshwater bacterium *Cellvibrio* sp. KY-GH-1 (KCTC13629BP) using only β -agarases.¹⁶³

3.1.2 Biodegradation of marine proteins

3.1.2.1 Collagen. The collagen molecule contains three peptide chains that are wound together to form a triple helix.

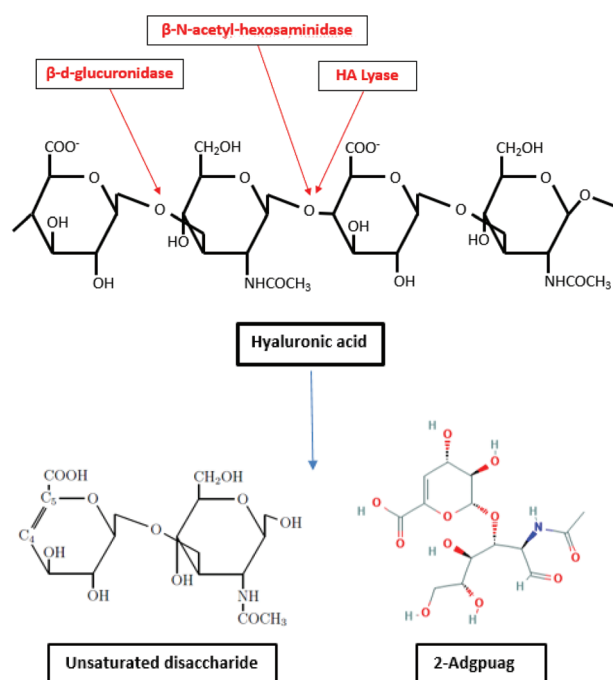


Fig. 6 Degradation of hyaluronic acid by enzymes β -D-glucuronidase, hyaluronate lyase and β -N-acetyl-hexosaminidase (inspired by ref. 157).

In nature, it is found as the insoluble polymerized form of fibers and filaments. The primary sequence of collagen comprises a repeating tripeptide (Gly-X-Y), in which X is proline and Y is hydroxyproline. The chains form a left-handed polyproline helix structure and three chains interact and adopt a right-handed triple helix.¹⁶⁴ Collagen molecules are mostly found in fibrous tissues in which each molecule is joined to the other in an end to end pattern.¹⁶⁵ Up to date, about 20

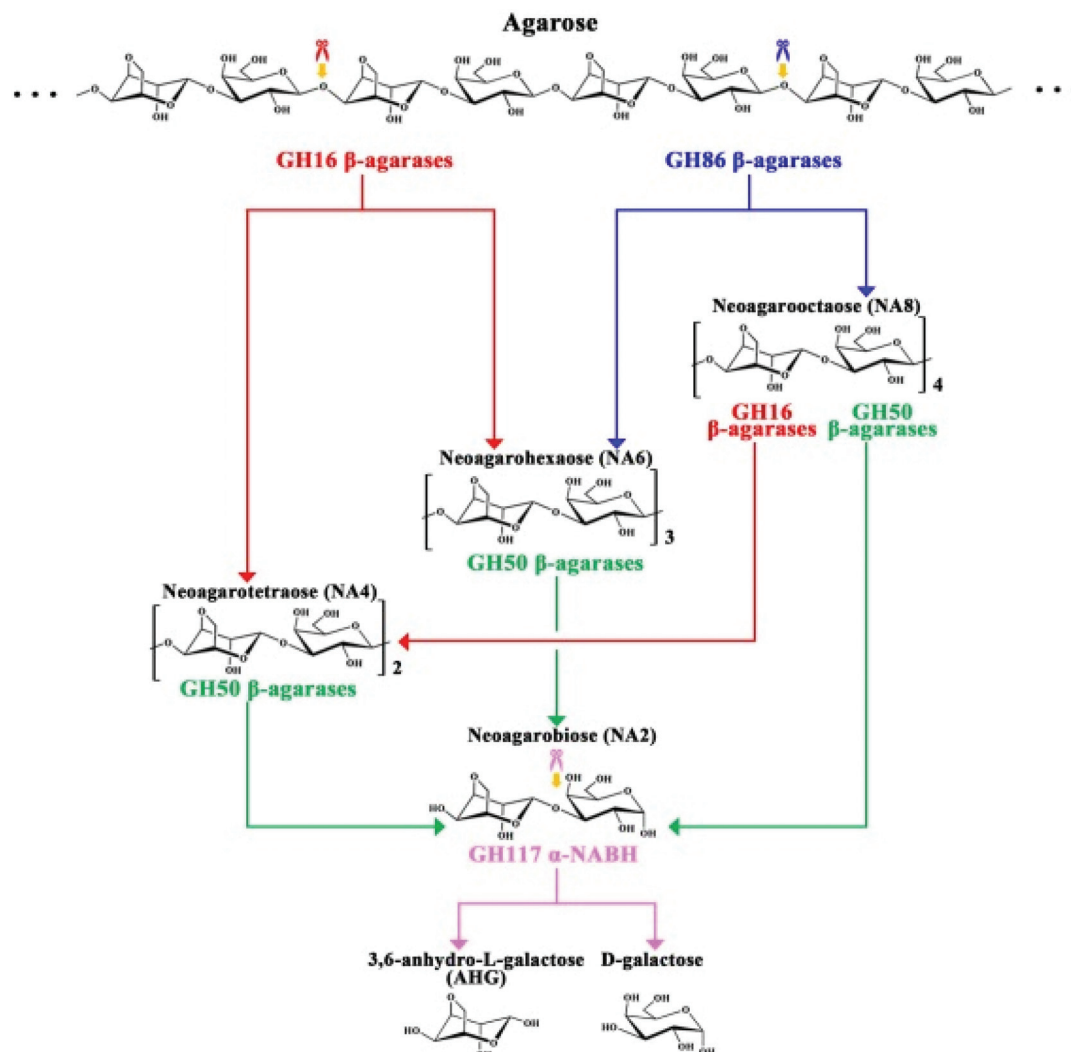


Fig. 7 Schematic diagram of the process by which *Cellvibrio* sp. KY-GH-1 degrades agarose. Reprinted with permission from ref. 163, copyright Elsevier, 2019.

different types of collagens have been found (type I, II, III, ...). In the marine environment, collagen can be found in fishes, marine sponges or jellyfish.¹⁶⁶ Due to its diverse properties, such as haemostatic effect, low antigenicity, and good mechanical characteristics, collagen has potential in biomedical and cosmetic sectors. Moreover, it is a promising candidate in tissue engineering⁴⁵ and cosmetics⁴⁶ because of its excellent biocompatibility and biodegradability.⁴⁷

Collagen is degraded by microorganisms that release collagenases, and some of those bacteria in the marine environment are *Pseudomonas* and *Pseudomonas marinoglutinosa* (Fig. 2 and Table 2).^{82,83} Collagenases are endopeptidases that can digest native collagen in the triple helix region. Collagenases catalyse the breakdown of collagen molecules to collagen fragments by breaking the peptide bonds (Fig. 8). Collagen fibrils (aggregates of collagen molecules) start degrading from the exterior. During the biodegradation

process, enzyme collagenase first binds to the triple helix at the outer surface to initiate the degradation. As the degradation proceeds, the enzyme gains access to the inner molecules resulting in the cracking of the triple helix. Further degradation of collagen molecules by the actions of other non-specific enzymes (like gelatinases and proteinases) that can cause cleavage of the primary protein fragments into small peptides and amino acids.¹⁶⁷

3.2 Biodegradation by oxidoreductase

Lytic polysaccharide monooxygenases (LPMOs, EC 1.14.99.54) belong to oxidoreductase (EC 1), and it is non-specific to the degradation of polysaccharides. LPMOs are found in fungi and bacteria, which catalyzes the cleavage of glycosidic bonds of polysaccharides by hydroxylation of one of the carbons in the bonds. LPMOs are mono-copper enzymes and the first LPMOs were discovered in the early 1990s but at the time they were

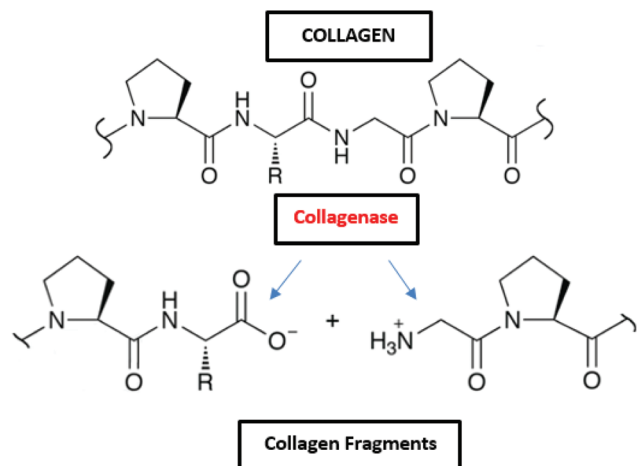


Fig. 8 Degradation of collagen by the enzyme collagenase.

considered as glycoside hydrolases that degrade cellulose. In 2011, they were named polysaccharide monooxygenases (PMOs) and then LPMOs.¹⁶⁸ The first fungal LPMO isolated was from *Talaromyces cellulolyticus* and was named as *TrCel61A* followed by another LPMO from *Trichoderma reesei* namely *TrCel61B*. Its function resulted in hydrolysis of cellulose but its crystal structure didn't support hydrolase characteristics.^{169,170} LPMOs have a flat surface on the overall structure with a putative metal-binding site. Its structure include a copper coordination sphere composed of three nitrogen ligands coordinated by two histidine residues,^{171,172} and its unique structure partially decides its function to oxidize polysaccharides.¹⁷³ LPMOs can break down the structure of polysaccharide chains in a crystalline environment. It also eases the access to the biopolymer for hydrolytic enzymes. LPMOs act on polysaccharides in different ways: either with a monooxygenase reaction or peroxygenase reaction (Fig. 9).¹⁷⁴ Both mechanisms lead to the hydroxylation of carbons C1 or C4 in the polysaccharides. Many LPMOs have been reported to work on polysaccharides like xylan, chitin, cellulose or starch.¹⁷³ Recently, Vaaje-Kolstad *et al.* showed that dioxygen is essential for the monooxygenase reaction and therefore, for the degradation of polysaccharides. Hydrogen peroxide is formed during the reaction because of the reduction of O₂ and because of the oxidase activity of LPMOs. After the reduction of O₂, H₂O₂ act as a co-substrate enabling further catalytic cycles of the enzyme.^{174,175} The reactions involved in the catalysis are shown in the Fig. 9(a) and (b) with the LPMOs from *Lentinus similis* (namely, *LsAA9A*).

Studies on the degradation of chitin by LPMOs have also been investigated. Kuusk *et al.* studied the kinetics of this reaction with the AA10 LPMO from *Serratia marcescens*, also known as SmLPMO10A¹⁷⁶ and proposed a reaction scheme for the degradation of chitin nanowhiskers. It seems that the degradation of chitin by LPMOs highly depend on the O₂, H₂O₂ and polysaccharide concentration. Only a few LPMOs from the

marine environment have been described, which include one LPMO in a shipworm symbiont: *Teredinibacter turnerae*.¹⁷⁷

4 Biodegradation of marine bio-based materials in marine ecosystem: where do we stand today?

Contrarily to the investigation on biodegradation of natural polymers, up to date, few studies have been done on the biodegradation of biopolymer-based materials. Some of these studies have been done in aquatic ecosystems. Brandl *et al.*¹⁷⁸ carried out the biodegradation experiments of polyhydroxyalkanoates (PHA) bottles in a Swiss lake, and Imam *et al.*¹⁷⁹ studied the biodegradation of starch-polyethylene films in a river. As for the marine environment, biodegradation studies have been made on films and bags containing starch.^{180,181} The biodegradation of polylactic acid (PLA)-based materials also had a few reports in seawater and marine ecosystems, which includes: PLA films^{118,182} as well as flax/PLA composites,¹⁸³ but they showed very little biodegradation in the marine environment.

Regarding the marine biopolymer-based materials, the degradation studies in the marine environment is scarcely found with exceptions on chitin and chitosan films. It is probably because that chitosan-based films for food packaging has been developed and ready for commercialization¹⁸⁴ like "The Shellworks" using chitin and "CuanTec" using chitosan. However, the degradation studies for chitin and chitosan films were mainly in soil environment with only a few in aquatic environments. The biodegradation rate of chitin films was higher than chitosan films using degrading enzymes from soil (*Sphingobacterium multivorum*).¹⁸⁵ Another biodegradation study on polyethylene (PE)-chitin and polyethylene-chitosan films in soil environment was reported with bacteria extracted from soil (*Serratia marcescens*, *Pseudomonas aeruginosa*),¹⁸⁶ and results showed that 100% of the chitin and chitosan films were degraded in soil environment after six months, for which rate of biodegradation was higher for PE-chitin and PE-chitosan films than starch-based films. Marine bacterium *Pseudoalteromonas* sp. was reported to colonize on chitin films and the bacterial chitinase was assessed¹⁸⁷ but no evaluation was found on the biodegradation of chitin films by such bacteria. There is one study reporting the biodegradability of polyhydroxybutyrate (PHB)-chitin and PHB-chitosan films in river water. The blended films degraded over 60% after 30 days showing a better biodegradability than films made of pure biopolymer.¹⁸⁸

Monitoring the biodegradation of materials in environmental conditions can be done in various ways. It can be done in laboratory with enzyme assays, plate tests, respiration tests. Still, the best measure of the fate of a material is by exposure in a marine environment. However, the results are relevant only to the specific environment it was exposed to. The material can be then analysed by different methods to assess

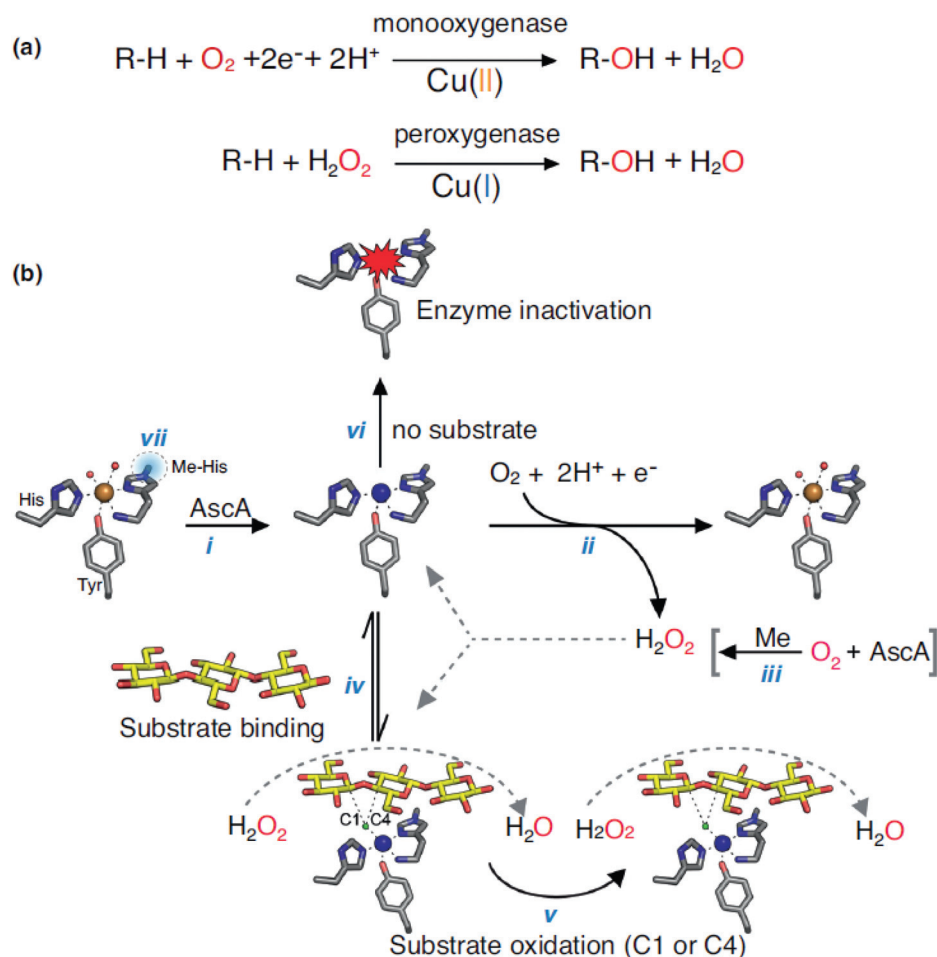


Fig. 9 (a) Monooxygenase and peroxygenase reactions occurring with LPMOs and (b) overview of LPMOs reactions (LsAA9A). Reprinted with permission from ref. 173 copyright Elsevier, 2019.

its degradation for instance: visual examination, infrared or ultraviolet spectroscopy, nuclear magnetic resonance, X-ray diffraction, etc.

As shown, in this section, there is a limited knowledge on the biodegradation mechanisms and experiments of biopolymer-based materials, including in marine environments. This shows the need for future research to determine and define methods to assess biodegradation in marine ecosystems, and to suggest novel directions on the development of sustainable bio-based materials and products and their rapid biodegradation.

5 Conclusion and future perspectives

The problematic biodegradation of petroleum-based polymers in the marine environment is a serious environmental issue that needs to be addressed. The natural polymers derived from marine sources represent an alternative to petroleum-based products as they are biodegradable and renewable.

In this sense, marine biopolymers have shown a great interest in many commercial applications in cosmetics, packaging, construction or medicine. Moreover, it is highly optimistic regarding the potential fast biodegradation of those marine biopolymers in the marine ecosystems by several microorganisms. To date, different biopolymers-degrading enzymes were identified in marine environment, including hydrolases, and lyases and LPMOs. Their catalysis mechanisms of marine polymers are well described; nevertheless, the number of isolated organisms and enzymes in this regard is limited across the scientific literature, suggesting that there are many to discover. Also, the elucidation of their biodegradation in marine ecosystem is necessary to encourage their use in future.

This review paper fills up a gap to describe systematically the biodegradability of must popular marine biopolymers in the marine environment. Nonetheless, it was observed that there is a limited knowledge on the biodegradation mechanisms of biopolymer-based materials, including in marine environments.

In this context, we would suggest some directions for future scientific research: (i) to isolate microorganisms from different

marine ecosystems (including, extreme environments) for the discovery of new biopolymer-degrading robust enzymes; (ii) to focus on biodegradation experiments of biopolymer-based materials and products; and (iii) to understand and describe the biodegradation mechanisms of biopolymer-based materials and products to accelerate their commercialization and use.

This review is a support for the development of sustainable marine environments and the green strategies to overcome marine environmental concerns.

Conflicts of interest

There are no conflicts to declare.

Acknowledgements

This work was carried under the framework of E2S UPPA Partnership Chair MANTA (Marine Materials) funded by the 'Investissements d'Avenir' French program managed by ANR, grant number #ANR-16-IDEX-0002; by the Région Nouvelle-Aquitaine and by the Communauté d'Agglomération du Pays Basque (France); and by the FLAG "Côte Basque-Sud Landes" through the community-led local development (CLDD: ref number # PFEA621219CR0720005 and ref number # PFEA621219CR0720024) program funded by the European Maritime and Fisheries Fund (EMFF) and the Région Nouvelle-Aquitaine, France. M. Thomas is the recipient of a Make Our Planet Great Again PhD Fellowship (grant number # MOPGA-927921L) co-funded by Campus France with the Ministère de l'Europe et des Affaires étrangères, and E2S UPPA.

References

- 1 G. Boeuf, *C. R. Biol.*, 2011, **334**, 435–440.
- 2 G. S. Dhillon, S. Kaur, S. K. Brar and M. Verma, *Crit. Rev. Biotechnol.*, 2013, **33**, 379–403.
- 3 A. Steinbüchel, *Curr. Opin. Biotechnol.*, 1992, **3**, 291–297.
- 4 M. Claverie, C. McReynolds, A. Petitpas, M. Thomas and S. C. M. Fernandes, *Polymers*, 2020, **12**, 1002.
- 5 A. A. Shah, F. Hasan, A. Hameed and S. Ahmed, *Biotechnol. Adv.*, 2008, **26**, 246–265.
- 6 R. Geyer, J. R. Jambeck and K. L. Law, *Sci. Adv.*, 2017, **3**, e1700782.
- 7 M. Roser, *Our World in Data*, 2018.
- 8 L. Lebreton, B. Slat, F. Ferrari, B. Sainte-Rose, J. Aitken, R. Marthouse, S. Hajbane, S. Cunsolo, A. Schwarz, A. Levivier, K. Noble, P. Debeljak, H. Maral, R. Schoeneich-Argent, R. Brambini and J. Reisser, *Sci. Rep.*, 2018, **8**, 4666.
- 9 J. A. Glaser, in *Plastics in the Environment*, ed. A. Gomiero, IntechOpen, 2019.
- 10 J.-D. Gu, *Int. Biodeterior. Biodegrad.*, 2003, **52**, 69–91.
- 11 K. Mattsson, L.-A. Hansson and T. Cedervall, *Environ. Sci.: Processes Impacts*, 2015, **17**, 1712–1721.
- 12 M. Smith, D. C. Love, C. M. Rochman and R. A. Neff, *Curr. Environ. Health Rep.*, 2018, **5**, 375–386.
- 13 C. J. Thiele, M. D. Hudson, A. E. Russell, M. Saluveer and G. Sidaoui-Haddad, *Sci. Rep.*, 2021, **11**, 2045.
- 14 S. Reynaud, A. Aynard, B. Grassl and J. Gigault, *Curr. Opin. Colloid Interface Sci.*, 2021, 101528.
- 15 R. Fernández-Marín, S. C. M. Fernandes, M. Á. A. Sánchez and J. Labidi, *Food Hydrocolloids*, 2022, **123**, 107119.
- 16 S. C. M. Fernandes, A. Alonso-Varona, T. Palomares, V. Zubillaga, J. Labidi and V. Bulone, *ACS Appl. Mater. Interfaces*, 2015, **7**, 16558–16564.
- 17 A. M. Salaberria, R. H. Diaz, J. Labidi and S. C. M. Fernandes, *React. Funct. Polym.*, 2015, **89**, 31–39.
- 18 S. Fernandes, C. S. R. Freire, C. P. Neto and A. Gandini, *Green Chem.*, 2008, **10**, 93–97.
- 19 S. C. M. Fernandes, C. S. R. Freire, A. J. D. Silvestre, J. Desbrières, A. Gandini and C. P. Neto, *Ind. Eng. Chem. Res.*, 2010, **49**, 6432–6438.
- 20 V. K. Thakur and S. I. Voicu, *Carbohydr. Polym.*, 2016, **146**, 148–165.
- 21 R. Ylitalo, S. Lehtinen, E. Wuolijoki, P. Ylitalo and T. Lehtimäki, *Arzneimittelforschung*, 2011, **52**, 1–7.
- 22 A. Muxika, I. Zugasti, P. Guerrero and K. de la Caba, in *Reference Module in Food Science*, Elsevier, 2017, p. B9780081005965224000.
- 23 R. Sharif, M. Mujtaba, M. Ur Rahman, A. Shalmani, H. Ahmad, T. Anwar, D. Tianchan and X. Wang, *Molecules*, 2018, **23**, 872.
- 24 S. C. M. Fernandes, C. S. R. Freire, A. J. D. Silvestre, C. P. Neto, A. Gandini, J. Desbrières, S. Blanc, R. A. S. Ferreira and L. D. Carlos, *Carbohydr. Polym.*, 2009, **78**, 760–766.
- 25 I. Aranaz, N. Acosta, C. Civera, B. Elorza, J. Mingo, C. Castro, M. Gandía and A. Heras Caballero, *Polymers*, 2018, **10**, 213.
- 26 S. M. Ahsan, M. Thomas, K. K. Reddy, S. G. Sooraparaju, A. Asthana and I. Bhatnagar, *Int. J. Biol. Macromol.*, 2018, **110**, 97–109.
- 27 M. Yadav, P. Goswami, K. Paritosh, M. Kumar, N. Pareek and V. Vivekanand, *Bioresour. Bioprocess.*, 2019, **6**, 8.
- 28 N. Morin-Crini, E. Lichtfouse, G. Torri and G. Crini, *Environ. Chem. Lett.*, 2019, **17**, 1667–1692.
- 29 A. Nagarajan, A. Shanmugam and A. Zackaria, *J. Algal Biomass Util.*, 2016, **7**, 45–55.
- 30 A. Dalmoro, A. A. Barba, G. Lamberti, M. Grassi and M. d'Amore, *Adv. Polym. Technol.*, 2012, **31**, 219–230.
- 31 H. H. Tønnesen and J. Karlsen, *Drug Dev. Ind. Pharm.*, 2002, **28**, 621–630.
- 32 R. Pereira, A. Carvalho, D. C. Vaz, M. H. Gil, A. Mendes and P. Bártolo, *Int. J. Biol. Macromol.*, 2013, **52**, 221–230.
- 33 E. Alsberg, K. W. Anderson, A. Albeiruti, R. T. Franceschi and D. J. Mooney, *J. Dent. Res.*, 2001, **80**, 2025–2029.
- 34 G. Kogan, L. Šoltés, R. Stern, J. Schiller and R. Mendichi, in *Studies in Natural Products Chemistry*, Elsevier, 2008, vol. 34, pp. 789–882.

- 35 A. Huynh and R. Priefer, *Carbohydr. Res.*, 2020, **489**, 107950.
- 36 C. Chircov, A. M. Grumezescu and L. E. Bejenaru, *Rom. J. Morphol. Embryol.*, 2018, **59**, 71–76.
- 37 N. J. Lowe, C. A. Maxwell, P. Lowe, M. G. Duickb and K. Shah, *J. Am. Acad. Dermatol.*, 2001, **45**, 930–933.
- 38 A. Sionkowska, B. Kaczmarek, M. Michalska, K. Lewandowska and S. Grabska, *Pure Appl. Chem.*, 2017, **89**, 1829–1839.
- 39 V. V. Menon, in *Handbook of Marine Macroalgae*, ed. S.-K. Kim, John Wiley & Sons, Ltd, Chichester, UK, 2011, pp. 541–555.
- 40 A. Mortensen, F. Aguilar, R. Crebelli, A. Di Domenico, M. J. Frutos, P. Galtier, D. Gott, U. Gundert-Remy, C. Lambré, J. Leblanc, O. Lindtner, P. Moldeus, P. Mosesso, A. Oskarsson, D. Parent-Massin, I. Stankovic, I. Waalkens-Berendsen, R. A. Woutersen, M. Wright, M. Younes, L. Brimer, P. Peters, J. Wiesner, A. Christodoulidou, F. Lodi, A. Tard and B. Dusemund, *EFSA J.*, 2016, **14**, 12.
- 41 M. Bonnet, J. C. Lagier, D. Raoult and S. Khelaifia, *New Microbes New Infect.*, 2020, **34**, 100622.
- 42 P. Y. Lee, J. Costumbrado, C.-Y. Hsu and Y. H. Kim, *JoVE*, 2012, 3923.
- 43 O. Coskun, *North. Clin. Istanbul*, 2016, **3**(2), 156–160.
- 44 V. Rudovica, A. Rotter, S. P. Gaudêncio, L. Novoveská, F. Akgül, L. K. Akslen-Hoel, D. A. M. Alexandrino, O. Anne, L. Arbidans, M. Atanassova, M. Beldowska, J. Beldowski, A. Bhatnagar, O. Bikovens, V. Bisters, M. F. Carvalho, T. S. Catalá, A. Dubnika, A. Erdoğan, L. Ferrans, B. Z. Haznedaroglu, R. H. Setyobudi, B. Graca, I. Grinfelde, W. Hogland, E. Ioannou, Y. Jani, M. Katarzytė, S. Kikionis, K. Klun, J. Kotta, M. Kriipsalu, J. Labidi, L. Lukić Bilela, M. Martínez-Sanz, J. Oliveira, R. Ozola-Davidane, J. Pilecka-Ulcugaceva, K. Pospiskova, C. Rebours, V. Roussis, A. López-Rubio, I. Safarik, F. Schmieder, K. Stankevica, T. Tamm, D. Tasdemir, C. Torres, G. C. Varese, Z. Vincevica-Gaile, I. Zekker and J. Burlakovs, *Front. Mar. Sci.*, 2021, **8**, 723333.
- 45 Y.-S. Lim, Y.-J. Ok, S.-Y. Hwang, J.-Y. Kwak and S. Yoon, *Mar. Drugs*, 2019, **17**, 467.
- 46 J. Rao, R. Pallela and G. Prakash, in *Marine Cosmeceuticals*, CRC Press, 2011, pp. 77–104.
- 47 E. Berteau, D. Ionita, M. Simoiu, M. Paraschiv, R. Tatia, A. Apatean, M. Sidoroff and L. Tcacenco, *Bull. Mater. Sci.*, 2016, **39**, 377–383.
- 48 Mintel Companies, 2021.
- 49 A. Rudin and P. Choi, in *The Elements of Polymer Science & Engineering*, Elsevier, 2013, pp. 521–535.
- 50 J. Brizga, K. Hubacek and K. Feng, *One Earth*, 2020, **3**, 45–53.
- 51 S. R. Unger, T. A. Hottle, S. R. Hobbs, C. L. Thiel, N. Champion, M. M. Bilec and A. E. Landis, *J. Health Serv. Res. Policy*, 2017, **22**, 218–225.
- 52 N. J. Bennett, A. M. Cisneros-Montemayor, J. Blythe, J. J. Silver, G. Singh, N. Andrews, A. Calò, P. Christie, A. Di Franco, E. M. Finkbeiner, S. Gelcich, P. Guidetti, S. Harper, N. Hotte, J. N. Kittinger, P. Le Billon, J. Lister, R. López de la Lama, E. McKinley, J. Scholtens, A.-M. Solàs, M. Sowman, N. Talloni-Álvarez, L. C. L. Teh, M. Voyer and U. R. Sumaila, *Nat. Sustainable*, 2019, **2**, 991–993.
- 53 D. Belhabib, P. Le Billon and N. J. Bennett, *Nat. Hum. Behav.*, 2022, **6**, 6–8.
- 54 M. U. Sheth, S. K. Kwartler, E. R. Schmaltz, S. M. Hoskinson, E. J. Martz, M. M. Dunphy-Daly, T. F. Schultz, A. J. Read, W. C. Eward and J. A. Somarelli, *Front. Mar. Sci.*, 2019, **6**, 624.
- 55 H. Sun, L. Gao, C. Xue and X. Mao, *Compr. Rev. Food Sci. Food Saf.*, 2020, **19**, 2767–2796.
- 56 G. Wang, D. Huang, J. Ji, C. Völker and F. R. Wurm, *Adv. Sci.*, 2021, **8**, 2001121.
- 57 P. Sharma, P. S. Slathia, N. Raina and D. Bhagat, in *Freshwater Microbiology*, Elsevier, 2019, pp. 341–392.
- 58 J. Klier, O. Dellwig, T. Leipe, K. Jürgens and D. P. R. Herlemann, *Front. Microbiol.*, 2018, **9**, 236.
- 59 E. D. Enger and B. F. Smith, *Environmental science: a study of interrelationships*, McGraw-Hill Education, New York, NY, Fifteenth edition, 2019.
- 60 S. Chakraborty, A. Khopade, C. Kokare, K. Mahadik and B. Chopade, *J. Mol. Catal. B: Enzym.*, 2009, **58**, 17–23.
- 61 Y. Ohta, Y. Hatada, M. Miyazaki, Y. Nogi, S. Ito and K. Horikoshi, *Curr. Microbiol.*, 2005, **50**, 212–216.
- 62 P. S. Meadows and J. I. Campbell, *An Introduction to Marine Science*, Springer Netherlands, Dordrecht, 1988.
- 63 A. Boetius, *Helgoländer Meeresuntersuchungen*, 1995, **49**, 177–187.
- 64 E. Krause, A. Wichels, L. Giménez, M. Lunau, M. B. Schilhabel and G. Gerdt, *PLoS One*, 2012, **7**, e47035.
- 65 S. Srimathi, G. Jayaraman, G. Feller, B. Danielsson and P. R. Narayanan, *Extremophiles*, 2007, **11**, 505–515.
- 66 K. Mukai, K. Yamada and Y. Doi, *Polym. Degrad. Stab.*, 1994, **43**, 319–327.
- 67 T. Ojima, in *Marine Enzymes for Biocatalysis*, Elsevier, 2013, pp. 333–371.
- 68 N. R. Nair, V. C. Sekhar, K. M. Nampoothiri and A. Pandey, in *Current Developments in Biotechnology and Bioengineering*, Elsevier, 2017, pp. 739–755.
- 69 T. Miyamoto, S. Takahashi, H. Ito, H. Inagaki and Y. Noishiki, *J. Biomed. Mater. Res.*, 1989, **23**, 125–133.
- 70 W. Liu, Y. Ma, L. Ai, W. Li, W. Li, H. Li, C. Zhou and B. Luo, *ACS Biomater. Sci. Eng.*, 2019, **5**, 5316–5326.
- 71 T. G. Volova, A. N. Boyandin, A. D. Vasiliev, V. A. Karpov, S. V. Prudnikova, O. V. Mishukova, U. A. Boyarskikh, M. L. Filipenko, V. P. Rudnev, B. Bá Xuân, V. Việt Dũng and I. I. Gitelson, *Polym. Degrad. Stab.*, 2010, **95**, 2350–2359.
- 72 J. P. Harrison, M. Sapp, M. Schratzberger and A. M. Osborn, *Mar. Technol. Soc. J.*, 2011, **45**, 12–20.
- 73 N. Mohanan, Z. Montazer, P. K. Sharma and D. B. Levin, *Front. Microbiol.*, 2020, **11**, 580709.

- 74 R. Osawa and T. Koga, *Lett. Appl. Microbiol.*, 1995, **21**, 288–291.
- 75 H. Yagi, A. Fujise, N. Itabashi and T. Ohshiro, *Biosci., Biotechnol., Biochem.*, 2016, **80**, 2338–2346.
- 76 Y. Zhu, L. Wu, Y. Chen, H. Ni, A. Xiao and H. Cai, *Microbiol. Res.*, 2016, **182**, 49–58.
- 77 J.-P. Chessa, G. Feller and C. Gerday, *Can. J. Microbiol.*, 1999, **45**, 452–457.
- 78 D. R. Humphry, A. George, G. W. Black and S. P. Cummings, *Int. J. Syst. Evol. Microbiol.*, 2001, **51**, 1235–1243.
- 79 R. Salas-Ovillo, D. Gálvez-López, A. Vázquez-Ovando, M. Salvador-Figueroa and R. Rosas-Quijano, *PeerJ*, 2019, **7**, e6102.
- 80 M. Pasqualetti, P. Barghini, V. Giovannini and M. Fenice, *Molecules*, 2019, **24**, 1880.
- 81 V. de M. Oliveira, C. R. D. Assis, P. N. Herculano, M. T. H. Cavalcanti, R. de S. Bezerra and A. L. F. Porto, *Bol. Inst. Pesca*, 2017, **43**, 52–64.
- 82 M. Gautam and W. Azmi, *J. Appl. Biotechnol. Rep.*, 2017, **4**, 558–565.
- 83 H. Kazunori, M. Taku, Y. Michio, T. Masaharu, T. Hikoji, M. Tetsuo and S. Jiro, *Agric. Biol. Chem.*, 1971, **35**, 1651–1659.
- 84 Y. Sugano, I. Terada, M. Arita, M. Noma and T. Matsumoto, *Appl. Environ. Microbiol.*, 1993, **59**, 1549–1554.
- 85 M. Duckworth and J. R. Turvey, *Biochem. J.*, 1969, **113**, 139–142.
- 86 P. Potin, C. Richard, C. Rochas and B. Kloareg, *Eur. J. Biochem.*, 1993, **214**, 599–607.
- 87 T. Araki, M. Hayakawa, Z. Lu, S. Karita and T. Morishita, *J. Mar. Biotechnol.*, 1998, **6**, 260–265.
- 88 Z. Hu, B.-K. Lin, Y. Xu, M. Q. Zhong and G.-M. Liu, *J. Appl. Microbiol.*, 2009, **106**, 181–190.
- 89 J. Wang, H. Mou, X. Jiang and H. Guan, *Appl. Microbiol. Biotechnol.*, 2006, **71**, 833–839.
- 90 J. Vera, R. Alvarez, E. Murano, J. C. Slebe and O. Leon, *Appl. Environ. Microbiol.*, 1998, **64**, 4378–4383.
- 91 L. M. Morrice, M. W. McLEAN, F. B. Williamson and W. F. Long, *Eur. J. Biochem.*, 1983, **135**, 553–558.
- 92 T. Aoki, T. Araki and M. Kitamikado, *Eur. J. Biochem.*, 1990, **187**, 461–465.
- 93 X. T. Fu, H. Lin and S. M. Kim, *Appl. Microbiol. Biotechnol.*, 2008, **78**, 265–273.
- 94 H. J. van der Meulen and W. Harder, *Antonie van Leeuwenhoek*, 1975, **41**, 431–447.
- 95 H. Li, Z. Chi, X. Wang and C. Ma, *J. Ocean Univ. China*, 2007, **6**, 60–65.
- 96 A. Bhattacharya and B. I. Pletschke, *Enzyme Microb. Technol.*, 2014, **55**, 159–169.
- 97 F. Pérez-Pomares, V. Bautista, J. Ferrer, C. Pire, F. C. Marhuenda-Egea and M. J. Bonete, *Extremophiles*, 2003, **7**, 299–306.
- 98 X. Wang, G. Kan, X. Ren, G. Yu, C. Shi, Q. Xie, H. Wen and M. Betenbaugh, *BioMed Res. Int.*, 2018, **2018**, 1–16.
- 99 A. N. M. Ramli, M. A. Azhar, M. S. Shamsir, A. Rabu, A. M. A. Murad, N. M. Mahadi and R. Md. Illias, *J. Mol. Model*, 2013, **19**, 3369–3383.
- 100 P. V. Vardhan Reddy, S. S. Shiva Nageswara Rao, M. S. Pratibha, B. Sailaja, B. Kavya, R. R. Manorama, S. M. Singh, T. N. Radha Srinivas and S. Shivaji, *Res. Microbiol.*, 2009, **160**, 538–546.
- 101 A. Del-Cid, P. Ubilla, M.-C. Ravanal, E. Medina, I. Vaca, G. Levicán, J. Eyzaguirre and R. Chávez, *Appl. Biochem. Biotechnol.*, 2014, **172**, 524–532.
- 102 S. Elleuche, F. M. Qoura, U. Lorenz, T. Rehn, T. Brück and G. Antranikian, *J. Mol. Catal. B: Enzym.*, 2015, **116**, 70–77.
- 103 C. Bertoldo, F. Duffner, P. L. Jorgensen and G. Antranikian, *Appl. Environ. Microbiol.*, 1999, **65**, 2084–2091.
- 104 S. Violot, N. Aghajari, M. Czjzek, G. Feller, G. K. Sonan, P. Gouet, C. Gerday, R. Haser and V. Receveur-Bréchot, *J. Mol. Biol.*, 2005, **348**, 1211–1224.
- 105 C. Zhang and S.-K. Kim, *Mar. Drugs*, 2010, **8**, 1920–1934.
- 106 V. M. Pathak and Navneet, *Bioresour. Bioprocess.*, 2017, **4**, 15.
- 107 H. J. Hueck, *Int. Biodeterior. Biodegrad.*, 2001, **48**, 5–11.
- 108 J. H. Walsh, *Int. Biodeterior. Biodegrad.*, 2001, **48**, 16–25.
- 109 S. Wallström, E. Strömberg and S. Karlsson, *Polym. Test.*, 2005, **24**, 557–563.
- 110 F. Cappitelli, P. Principi and C. Sorlini, *Trends Biotechnol.*, 2006, **24**, 350–354.
- 111 S. Bonhomme, A. Cueur, A.-M. Delort, J. Lemaire, M. Sancelme and G. Scott, *Polym. Degrad. Stab.*, 2003, **81**, 441–452.
- 112 J.-P. Regnault, *Microbiologie générale*, Décarie, Vigot, Montréal; Paris (France), 1990.
- 113 C. A. Crispim and C. C. Gaylarde, *Microb. Ecol.*, 2005, **49**, 1–9.
- 114 J. Pelmont, *Biodégradations et métabolismes: les bactéries pour les technologies de l'environnement (Collection Grenoble sciences)*, EDP Sciences, 2005.
- 115 M. Shimao, *Curr. Opin. Biotechnol.*, 2001, **12**, 242–247.
- 116 A. Lugauskas, L. Levinskait and D. Pečiulyt, *Int. Biodeterior. Biodegrad.*, 2003, **52**, 233–242.
- 117 J. S. Webb, M. Nixon, I. M. Eastwood, M. Greenhalgh, G. D. Robson and P. S. Handley, *Appl. Environ. Microbiol.*, 2000, **66**, 3194–3200.
- 118 H. Tsuji and K. Suzuyoshi, *Polym. Degrad. Stab.*, 2002, **75**, 347–355.
- 119 N. Lucas, C. Bienaime, C. Belloy, M. Queneudec, F. Silvestre and J.-E. Nava-Saucedo, *Chemosphere*, 2008, **73**, 429–442.
- 120 Y. Li, S. Y. Park and J. Zhu, *Renewable Sustainable Energy Rev.*, 2011, **15**, 821–826.
- 121 *Biological wastewater treatment: principles, modelling and design*, ed. M. Henze, IWA Pub, London, 2008.
- 122 J.-B. Guillerme, C. Couteau and L. Coiffard, *Cosmetics*, 2017, **4**, 35.
- 123 D. Ozdil and H. M. Aydin, *J. Chem. Technol. Biotechnol.*, 2014, **89**, 1793–1810.

- 124 X. Hu, Y. Du, Y. Tang, Q. Wang, T. Feng, J. Yang and J. F. Kennedy, *Carbohydr. Polym.*, 2007, **70**, 451–458.
- 125 P. K. Dutta, M. N. V. Ravikumar and J. Dutta, *J. Macromol. Sci., Part C: Polym. Rev.*, 2002, **42**, 307–354.
- 126 W. Herth, A. Kuppel and E. Schnepf, *J. Cell Biol.*, 1977, **73**, 311–321.
- 127 W. Herth, *J. Ultrastruct. Res.*, 1979, **68**, 16–27.
- 128 R. A. Smucker, *Biochem. Syst. Ecol.*, 1991, **19**, 357–369.
- 129 V. Zargar, M. Asghari and A. Dashti, *ChemBioEng Rev.*, 2015, **2**, 204–226.
- 130 B. Bradić, U. Novak and B. Likozar, *Green Process. Synth.*, 2019, **9**, 13–25.
- 131 A. M. Salaberria, S. C. M. Fernandes, R. H. Diaz and J. Labidi, *Carbohydr. Polym.*, 2015, **116**, 286–291.
- 132 A. Labidi, A. M. Salaberria, S. C. M. Fernandes, J. Labidi and M. Abderrabba, *J. Dispersion Sci. Technol.*, 2020, **41**, 828–840.
- 133 A. Labidi, A. Salaberria, S. Fernandes, J. Labidi and M. Abderrabba, *Materials*, 2019, **12**, 361.
- 134 A. Labidi, A. M. Salaberria, S. C. M. Fernandes, J. Labidi and M. Abderrabba, *J. Taiwan Inst. Chem. Eng.*, 2016, **65**, 140–148.
- 135 A. M. Salaberria, J. Labidi and S. C. M. Fernandes, *Eur. Polym. J.*, 2015, **68**, 503–515.
- 136 A. M. Salaberria, J. Labidi and S. C. M. Fernandes, *Chem. Eng. J.*, 2014, **256**, 356–364.
- 137 A. Salaberria, R. H. Diaz, M. Andrés, S. Fernandes and J. Labidi, *Materials*, 2017, **10**, 546.
- 138 A. M. Salaberria, R. Teruel-Juanes, J. D. Badia, S. C. M. Fernandes, V. Sáenz de Juano-Arbona, J. Labidi and A. Ribes-Greus, *Compos. Sci. Technol.*, 2018, **167**, 323–330.
- 139 R. Fernández-Marín, J. Labidi, M. Á. Andrés and S. C. M. Fernandes, *Polym. Degrad. Stab.*, 2020, 109227.
- 140 V. Zubillaga, A. M. Salaberria, T. Palomares, A. Alonso-Varona, S. Kootala, J. Labidi and S. C. M. Fernandes, *Biomacromolecules*, 2018, **19**, 3000–3012.
- 141 V. Zubillaga, A. Alonso-Varona, S. C. M. Fernandes, A. M. Salaberria and T. Palomares, *Int. J. Mol. Sci.*, 2020, **21**, 1004.
- 142 *Nitrogen-Containing Macromolecules in the Bio- and Geosphere*, ed. B. A. Stankiewicz and P. F. van Bergen, American Chemical Society, Washington, DC, 1998, vol. 707.
- 143 L. Ralph Berger and D. M. Reynold, *Biochim. Biophys. Acta*, 1958, **29**, 522–534.
- 144 *Chitin in Nature and Technology*, ed. R. Muzzarelli, C. Jeuniaux and G. W. Gooday, Springer US, Boston, MA, 1986.
- 145 S. Beier and S. Bertilsson, *Front. Microbiol.*, 2013, **4**, 149.
- 146 Q. Yan and S. S. Fong, *Bioresour. Bioprocess.*, 2015, **2**, 31.
- 147 A. Sørbotten, S. J. Horn, V. G. H. Eijsink and K. M. Vårum, *FEBS J.*, 2005, **272**, 538–549.
- 148 K.-E. L. Eriksson, R. A. Blanchette and P. Ander, *Microbial and Enzymatic Degradation of Wood and Wood Components*, Springer Berlin Heidelberg, Berlin, Heidelberg, 1990.
- 149 C. Ma, X. Li, K. Yang and S. Li, *Mar. Drugs*, 2020, **18**, 126.
- 150 Y. Zhou, X. Chen, X. Li, Y. Han, Y. Wang, R. Yao and S. Li, *Molecules*, 2019, **24**, 183.
- 151 K. Y. Lee and D. J. Mooney, *Prog. Polym. Sci.*, 2012, **37**, 106–126.
- 152 I. A. Brownlee, A. Allen, J. P. Pearson, P. W. Dettmar, M. E. Havler, M. R. Atherton and E. Onsøyen, *Crit. Rev. Food Sci. Nutr.*, 2005, **45**, 497–510.
- 153 J.-C. Tang, H. Taniguchi, H. Chu, Q. Zhou and S. Nagata, *Lett. Appl. Microbiol.*, 2009, **48**, 38–43.
- 154 K. Schaumann and G. Weide, in *Thirteenth International Seaweed Symposium*, ed. S. C. Lindstrom and P. W. Gabrielson, Springer Netherlands, Dordrecht, 1990, pp. 589–596.
- 155 M. Cardoso, R. Costa and J. Mano, *Mar. Drugs*, 2016, **14**, 34.
- 156 R. A. Joy, N. Vikkath and P. S. Ariyannur, *Drug Metab. Pers. Ther.*, 2018, **33**, 15–32.
- 157 Y. Dongol, B. L. Dhananjaya, R. K. Shrestha and G. Aryal, in *Pharmacology and Therapeutics*, ed. S. J. T. Gowder, InTech, 2014.
- 158 J. Necas, L. Bartosikova, P. Brauner and J. Kolar, *Vet. Med.*, 2008, **53**, 397–411.
- 159 M. J. Jedrzejewski, L. V. Mello, B. L. de Groot and S. Li, *J. Biol. Chem.*, 2002, **277**, 28287–28297.
- 160 W. Han, W. Wang, M. Zhao, K. Sugahara and F. Li, *J. Biol. Chem.*, 2014, **289**, 27886–27898.
- 161 S. H. Knutsen, D. E. Myslabodski, B. Larsen and A. I. Usov, *Bot. Mar.*, 1994, **37**(2), 163–170.
- 162 W.-J. Chi, Y.-K. Chang and S.-K. Hong, *Appl. Microbiol. Biotechnol.*, 2012, **94**, 917–930.
- 163 G. H. Kwon, M. J. Kwon, J. E. Park and Y. H. Kim, *Biotechnol. Rep.*, 2019, **23**, e00346.
- 164 H. P. Wiesmann, U. Meyer, U. Plate and H. J. Höhling, in *International Review of Cytology*, Elsevier, 2004, vol. 242, pp. 121–156.
- 165 R. G. Paul and A. J. Bailey, *Sci. World J.*, 2003, **3**, 138–155.
- 166 T. Silva, J. Moreira-Silva, A. Marques, A. Domingues, Y. Bayon and R. Reis, *Mar. Drugs*, 2014, **12**, 5881–5901.
- 167 W. Wagermaier and P. Fratzl, in *Polymer science: A comprehensive Reference*, Elsevier, 2012, pp. 35–55.
- 168 A. Levasseur, E. Drula, V. Lombard, P. M. Coutinho and B. Henrissat, *Biotechnol. Biofuels*, 2013, **6**, 41.
- 169 J. Karlsson, M. Saloheimo, M. Siika-aho, M. Tenkanen, M. Penttilä and F. Tjerneld, *Eur. J. Biochem.*, 2001, **268**, 6498–6507.
- 170 S. Karkehabadi, H. Hansson, S. Kim, K. Piens, C. Mitchinson and M. Sandgren, *J. Mol. Biol.*, 2008, **383**, 144–154.
- 171 W. T. Beeson, V. V. Vu, E. A. Span, C. M. Phillips and M. A. Marletta, *Annu. Rev. Biochem.*, 2015, **84**, 923–946.
- 172 R. J. Quinlan, M. D. Sweeney, L. Lo Leggio, H. Otten, J.-C. N. Poulsen, K. S. Johansen, K. B. R. M. Krogh, C. I. Jorgensen, M. Tovborg, A. Anthonsen, T. Tryfona, C. P. Walter, P. Dupree, F. Xu, G. J. Davies and P. H. Walton, *Proc. Natl. Acad. Sci. U. S. A.*, 2011, **108**, 15079–15084.

- 173 Z. Forsberg, M. Sørle, D. Petrović, G. Courtade, F. L. Aachmann, G. Vaaje-Kolstad, B. Bissaro, Å. K. Røhr and V. G. Eijsink, *Curr. Opin. Struct. Biol.*, 2019, **59**, 54–64.
- 174 B. Bissaro, Å. K. Røhr, G. Müller, P. Chylenski, M. Skaugen, Z. Forsberg, S. J. Horn, G. Vaaje-Kolstad and V. G. H. Eijsink, *Nat. Chem. Biol.*, 2017, **13**, 1123–1128.
- 175 G. Vaaje-Kolstad, B. Westereng, S. J. Horn, Z. Liu, H. Zhai, M. Sørle and V. G. H. Eijsink, *Science*, 2010, **330**, 219–222.
- 176 S. Kuusk, R. Kont, P. Kuusk, A. Heering, M. Sørle, B. Bissaro, V. G. H. Eijsink and P. Våljamäe, *J. Biol. Chem.*, 2019, **294**, 1516–1528.
- 177 C. A. Fowler, F. Sabbadin, L. Ciano, G. R. Hemsworth, L. Elias, N. Bruce, S. McQueen-Mason, G. J. Davies and P. H. Walton, *Biotechnol. Biofuels*, 2019, **12**, 232.
- 178 H. Brandl and P. Püchner, *Biodegradation*, 1992, **2**, 237–243.
- 179 S. H. Imam, J. M. Gould, S. H. Gordon, M. P. Kinney, A. M. Ramsey and T. R. Tosteson, *Curr. Microbiol.*, 1992, **25**, 1–8.
- 180 M. Rutkowska, A. Heimowska, K. Krasowska and H. Zofia Janik, *Pol. J. Environ. Stud.*, 2002, **11**, 267–274.
- 181 N.-C. Pauli, J. S. Petermann, C. Lott and M. Weber, *R. Soc. Open Sci.*, 2017, **4**, 170549.
- 182 H. Tsuji and K. Suzuyoshi, *Polym. Degrad. Stab.*, 2002, **75**, 357–365.
- 183 A. Le Duigou, P. Davies and C. Baley, *Polym. Degrad. Stab.*, 2009, **94**, 1151–1162.
- 184 R. Priyadarshi and J.-W. Rhim, *Innovative Food Sci. Emerging Technol.*, 2020, **62**, 102346.
- 185 T. NAKASHIMA, Y. NAKANO, Y. BIN and M. MATSUO, *J. Home Econ. Jpn.*, 2005, **56**, 889–897.
- 186 I. Makarios-Laham and T.-C. Lee, *J. Environ. Polym. Degrad.*, 1995, **3**, 31–36.
- 187 A. M. Baty, C. C. Eastburn, Z. Diwu, S. Techkarnjanaruk, A. E. Goodman and G. G. Geesey, *Appl. Environ. Microbiol.*, 2000, **66**, 3566–3573.
- 188 T. Ikejima and Y. Inoue, *Carbohydr. Polym.*, 2000, **41**, 351–356.