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ORIGINAL PAPER

Whole-cell electric sensor for determination of sodium dodecyl sulfate

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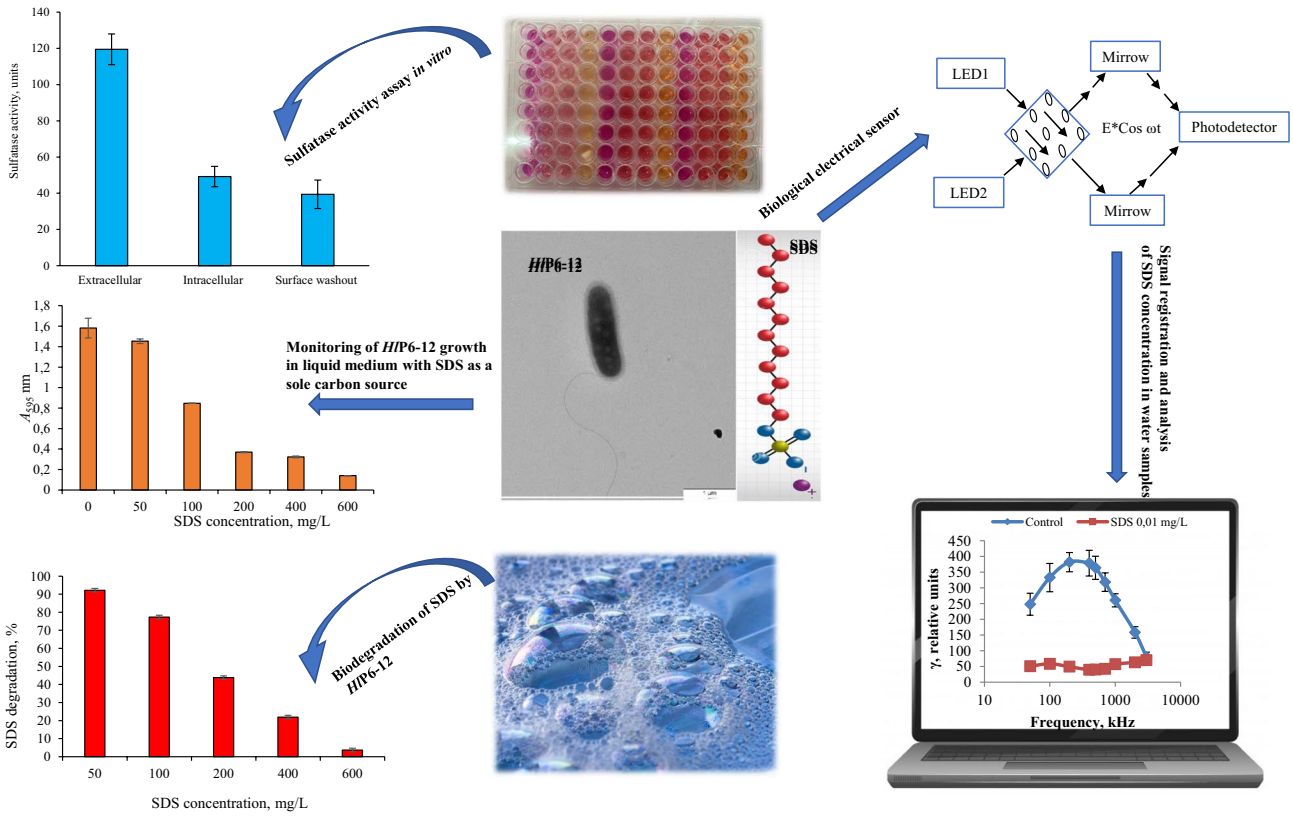
Abstract

Linear alkyl sulfates are a major class of surfactants that have large-scale industrial application and thus wide environmental release. These organic pollutants threaten aquatic environments and other environmental compartments. We show the promise of the use of a whole-cell electric sensor in the analysis of low or residual concentrations of sodium dodecyl sulfate (SDS) in aqueous solutions. On the basis of bioinformatic analysis and alkylsulfatase activity determinations, we chose the gram-negative bacterium *Herbaspirillum lusitanum*, strain P6–12, as the sensing element. Strain P6–12 could utilize 0.01–400 mg/L of SDS as a growth substrate. The electric polarizability of cell suspensions changed at all frequencies used (50–3000 kHz). The determination limit of 0.01 mg/L is much lower than the official requirements for the content of SDS in potable and process water (0.5 and 1.0 mg/L, respectively), and the analysis takes about 1–5 min. The promise of *H. lusitanum* P6–12 for use in the remediation of SDS-polluted soils is discussed.

Graphical abstract



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Keywords Alkylsulfatase · Biodegradation · Detection · *Herbaspirillum* · Sodium dodecyl sulfate · Whole-cell electric sensor

Introduction

Sodium dodecyl sulfate [$CH_3(CH_2)_{10}CH_2OSO_3Na$; SDS, E487; synonym: sodium lauryl sulfate, SLS] is a highly effective synthetic surfactant used in almost all everyday products, including soap, shampoo, toothpaste, detergent, and cleaner (Smulders et al. 2007; Devesa-Rey et al. 2011) as well as in various industries and in basic research (Adac et al. 2005; Liwarska and Bizukojc 2006; Martinez and Munoz 2007). SDS is also used as an adjuvant to improve the repellent effects, dispersion, and penetration of pesticides (Liwarska and Bizukojc 2006; Martinez and Munoz 2007) and as an emulsifying and bacteriostatic agent in pharmacology and medicine. In the EU countries, SDS is classed with the nonparenteral drugs (Liwarska and Bizukojc 2006; Martinez and Munoz 2007; EMA/CHMP/351898/2014). Owing to its ability to solubilize the virus envelope and destroy the capsid proteins, SDS is a potential inhibitor of enveloped (HIV, herpes simplex virus) and nonenveloped (human papilloma virus, rotavirus, poliovirus) viruses (Piret et al. 2002; Zhernov and Khaitov 2019).

In 2020, the SDS market volume was worth more than $590 million. It is expected to increase at a rate of 3.8% from 2021 to 2027 owing to the rapidly growing demand for cleaning, detergent, and personal care products (Pulidindi and Bhalerao 2021). The increased use of SDS and the imperfect treatment of industrial wastewater have led to large-scale pollution of the environment by SDS and its derivatives (Pettersson et al. 2000). According to EC Regulation No. 1272/2008, SDS (classified as Category 3) is moderately toxic, has acute toxic effects, and is a long-term hazard to water bodies. Surfactant accumulation in animals and humans leads to genetic disorders, malignant neoplasms, and immune response disorders (Martinez and Munoz 2007; Rosety et al. 2001; Forni et al. 2008; Messina et al. 2014).

The content of surfactants in environmental constituents is under the close attention of ecologists and is subject to mandatory monitoring. The limits to the content of SDS in potable and process water are 0.5 and 1.0 mg/L, respectively (Rao 1995; European Chemicals Agency [ECHA], 2008; ECHA REACH database: http://echa.europa.eu/information-on-chemicals/registered-substances).

An urgent need exists for specific and rapid methods to monitor SDS in water bodies and in industrial wastewater. The analytical methods used for SDS determination include capillary electrophoresis/mass spectrometry, turbidimetry, polarography, spectrophotometry and amperometry (Hayashi 1975; Petrovic and Barcelo 2000; Attaran et al. 2009; Taranova et al. 2002; Dey et al. 2020). These have several limitations, as they require expensive, high-tech equipment and are time-consuming and poorly sensitive (Hayashi 1975; Petrovic and Barcelo 2000). An effective alternative is the use of bacteria-based biosensors. Whole-cell biosensors have found application in environmental monitoring (Belkin 2003), pharmacology, and drug screening (Zager et al. 2010; Hillger et al. 2015). They have also been used to rapidly measure biochemical oxygen consumption in aquatic environments (Raudkivi et al. 2008; Abrevayaet et al. 2014). The integrity of the bacterial cell membrane after contact with a detergent and the induction of enzymatic catalysis, accompanied by redistribution of charges within the cell, can be recorded with an optical sensor (Bartnik 1992; Bunin and Voloshin 1996; Guliy et al. 2008; Guliy, Bunin 2020). However, the development of a unified device for surfactant analysis is a challenging task that requires the selection of promising prokaryotes (Cain 1994; van Ginkel 1996; Caracciolo et al. 2017).

The goal of this work was to select bacteria capable to utilizing SDS as the sole carbon source and to develop a bacteria-based sensor system for the determination of SDS in aqueous solutions.

Materials and methods

Bacteria and culture conditions

Herbaspirillum lusitanum P6–12 was from the IBPPM RAS Collection of Rhizosphere Microorganisms (IBPPM 515; http://collection.ibppm.ru). Cells were grown in a vitamin-supplemented liquid synthetic medium (Smol'kina et al. 2012) at 30 ± 1 °C for 22–24 h (until the end of the exponential growth phase).

Bioinformatics analysis

Search for amino acid sequences of *Herbaspirillum* sp. was carried out in the Protein NCBI and UniProt databases, annotated *Pseudomonas putida* alkyl sulfatase was chosen as a reference. Protein amino acid sequences were compared using the Protein BLAST tool (https://blast.ncbi.nlm.nih.gov). Search parameters: database—non-redundant protein sequences, organism—*Herbaspirillum* (taxid:963), blastp algorithm (protein–protein BLAST). The phylogenetic tree of amino acid sequences was built using the nearest neighbor joining algorithm (Saitou and Nei 1987.) using the MEGA 10 program (Kumar et al. 2018). Alignment of amino acid sequences was performed

using the MUSCLE method (Edgar 2004). The evolutionary distances were computed using the Poisson correction method (Zuckerkandl and Pauling 1965.) and are in the units of the number of amino acid substitutions per site. This analysis involved 6 amino acid sequences. All ambiguous positions were removed for each sequence pair (pairwise deletion option).

SDS degradation study by the methylene blue active substance (MBAS) method

The degradation experiment was conducted in 250-mL Erlenmeyer flasks containing 100 mL of a liquid medium composed as follows (g/L): K_2HPO_4, 0.25; $MgSO_4 \times 7H_2O$, 0.2; NaCl, 0.1; $Na_2MoO_4 \times 2H_2O$, 0.001; $MnSO_4 \times H_2O$, 0.002; $FeSO_4 \times 7H_2O$, 0.01; $CaCl_2 \times 2H_2O$, 0.02; $(NH_4)_2SO_4$, 1.0. Filter-sterilized SDS [Sigma–Aldrich, USA, L5750; 92.5–100.5% (on the basis of total alkyl sulfate content)] was used as the sole carbon source, and the concentration was increased progressively from 0 to 600 mg/L. The pH was adjusted to 7.2–7.4. After the medium was sterilized, it was inoculated with a suitable dilution of an overnight bacterial culture at $A_{600} = 0.1$ (10^9 CFU/mL). The degradation experiment was run in a shaking incubator at 200 rpm at 30 ± 1 °C. The incubation was done in triplicate, with appropriate duplicate controls (approximate duration, 24 h). Biomass increase was monitored by A_{595} measurements. Cultures grown in the liquid synthetic medium (Smol'kina et al. 2012) without SDS were used as a growth control.

SDS degradation was monitored by measuring the intensity of methylene blue in a chloroform extraction process [methylene blue active substance (MBAS) assay; Shukor et al. 2009). At time intervals, a sample of 5 mL was drawn from the culture medium. One hundred µL of the culture was added to 100 mL of a separating funnel containing 9.9 mL of deionized water; this was followed by the addition of 2.5 mL of a methylene blue solution and 1 mL of chloroform. The funnel was shaken vigorously for 15 s, and the mixture was left to separate and settle. The chloroform layer was drawn off into a second funnel. The extraction was repeated three times, with 1 mL of chloroform used each time. All chloroform extracts were combined in the second funnel before 5.0 mL of a washing solution was added. The funnel was then shaken vigorously for 15 s. The chloroform layer was drawn off into a volumetric flask. The washing solution was extracted twice with 1 mL of chloroform. All extracts were combined and diluted to 10 mL with chloroform. The absorbance was read at 652 nm against blank chloroform in a quartz cuvette. Calibration curves were generated by using standard solutions based on the weight of pure SDS. All experiments were run in triplicate, and results were expressed as mean ± SD.

Alkylsulfatase assay

Alkylsulfatase activity was measured by the rate of phenol red oxidation at 557 nm ($\varepsilon_{557} = 44{,}100\ M^{-1}\ cm^{-1}$) (Liang et al. 2014). The measurements were made in 96-well plates, and the absorbance decrease was read on a Spark 10 M tablet spectrophotometer (Tecan, Switzerland). The reaction mixture contained 0.2 mM HEPES (pH 7.5), 100 mM phenol red, 0.02% (wt/vol) SDS, and 20 µL sample. The reaction mixture was incubated at 37 °C for 60 min. One unit of activity is the amount of enzyme catalyzing the conversion of 1 µM substrate per min.

Bacterial cells were sedimented by centrifugation (10,000×*g*, 15 min), and the supernatant liquid was used to measure extracellular enzymatic activity.

Fractions of the cell surface enzymes were obtained with the cell shearing method of Eshdat et al. (1978), modified as follows: The cell suspension was passed several times through a syringe with a needle size of 0.8 × 38 mm. Treatment with 2-nitro-5-thiocyanobenzoic acid was omitted to avoid loss of enzyme activity. Then, the suspension was centrifuged (10,000×*g*, 10 min), and the supernatant liquid was separated from the sediment.

For intracellular enzymes, the cell suspension in phosphate-buffered saline (pH 7.2) was treated with ultrasound (37 kHz, 5 min) and then centrifuged (12,000×*g*, 15 min). The supernatant liquid was separated from the sediment and was filtered through a polytetrafluoroethylene (PTFE) filter (Merck).

Analysis with an electric sensor

To investigate the response of cells to SDS by measuring their electric polarizability, we used a sensing system based on the ELUS device (EloSystems GbR, Germany). Before analysis, the cells were washed free of the culture medium three times by centrifugation (2800×*g*, 5 min) in distilled water (electric conductivity, 1.6–1.8 µS/cm) and then they were centrifuged once (1000×*g*, 1 min) to remove cellular aggregates. Conductivity was measured with an HI 8733 m (HANNA, USA). The absorbance of the supernatant liquid was adjusted with distilled water to $A_{670} \sim 0.42$–0.45 (1.2×10^9 cells/mL of *H. lusitanum* P6–12). The measuring conditions were as follows: electric field strength, 89.4 V/cm; light wavelength, 670 nm (relative to vacuum); field application time, 4.5 s; volume of the measuring cell, ~ 1 mL; frequencies, 50, 100, 200, 400, 500, 700, 1000, 2000, and 3000 kHz. The method used to measure electric polarizability is based on the effect of an external electric field on cells suspended in an aqueous medium. The electric field gives rise to induced charges on the suspended particles. The structural mechanism of polarizability is characterized by the appearance of bulk charges at the

interfaces between adjacent media with different dielectric permittivities (the double electric layer–cell wall interface, the cell wall–cytoplasmic membrane interface, the cytoplasmic membrane–cytoplasm interface, etc.). The magnitude of the charges induced at the interfaces is proportional to the electric field strength E and depends on the ratio between the dielectric permittivities of the structures forming these interfaces. The phenomenological variable describing this effect is the particle polarizability tensor γ (Bottcher 1982); changes in this parameter were recorded in this work. Figure 1A shows the general scheme for the electric sensor.

The electrical sensory system for SDS determination is based on the effects of an electric field force on cells suspended in an aqueous medium, i.e. the method is based on using the effect of polarizability of particles in an electric field and measuring the optical manifestation of the cells polarization (Kerr effect/electro-optical effect). When an electric field is applied to a cell suspension, polarization of cellular structures occurs, as a result of which the cells acquire an induced cellular dipole moment. The physical manifestations of this effect is the electrical orientation of cells, which displays itself in the transition of cells to an oriented state. The phenomenon of electroorientation is due to the influence of an external electric field on the dipole moment of cells. Since all substances, including biological ones, contain free and bound charges, when an electric field is applied, two types of processes are observed in a substance. One of them—the movement (drift) of free charges (electrons and ions) through the thickness of the substance from one electrode to another is a conduction current. Another process is that the bound charges under the action of an external field are displaced within some acceptable but restricted limits, causing displacement currents and the appearance of an induced electric moment, as shown in Fig. 1B. This phenomenon of electric polarization of matter arises when a field is applied and decreases after its removal not instantly, but after some finite time, called the relaxation time (Guliy and Bunin 2020).

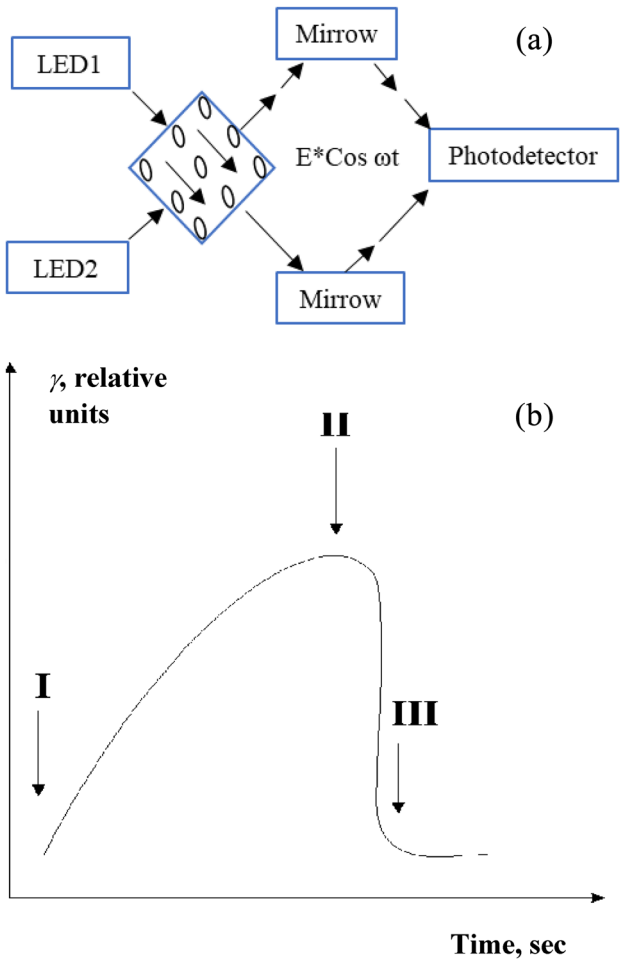

Fig. 1 **A** General scheme for the electric sensor and **B** Scheme of changes in the optical density of microbial cells under the orienting effect of an electric field for one of the mutually perpendicular directions of the optical beam: I—the moment the electric field is applied (chaotic orientation of the cells), II—the moment the field is turned off (the cells are in an oriented state), III—the moment the cells return to a state with a chaotic orientation

Because several interrelated variables (polarizability change, dielectric permittivity, optical signal change) are recorded simultaneously, the initial electric phenomenon can be measured only in relative units. The use of relative units simplifies the presentation of the experimental result because, by accepted international standards, relative units offer a way to calculate variables when the values of system quantities (voltage, current, resistance, power, etc.) are expressed as multipliers of a certain reference value taken as a unit.

For each set of experiments, at least five independent replicates were done. In the analysis of the electric polarizability, at least three replicate measurements were made for each datum point. Data were analyzed with Excel 2016 software (Microsoft Corp., USA). Confidence intervals were calculated for 95% confidence.

Results

Alkylsulfatase assay

Using a bioinformatics search, 5 amino acid sequences of *Herbaspirillum* alkylsulfatases were selected with a high level of homology to the reference alkylsulfatase (E-value less than 1e–130) (Table 1). The phylogenetic tree constructed based on the alignment of amino acid sequences is shown in the Fig. 2. The bioinformatic data were confirmed by a series of experiments to measure the alkylsulfatase activity of *H. lusitanum* P6–12 grown in the liquid medium

Table 1 Alkylsulfatases of *Herbaspirillum* spp. with high homology (score and *E*-value measures) to the reference alkylsulfatase of *P. putida*

Reference alkylsulfatase (protein ID)	*Herbaspirillum* alkylsulfatase (protein ID)	Score	*E*-value	Percent identity (%)
P. putida, Q9WWU5.1	*H. frisingense,* KAF1038947.1	413	2e–145	69.28
	H. huttiense, MBM7749534.1	407	3e–143	68.06
	H. hiltneri, WP _053198152.1	404	4e–142	69.40
	H. lusitanum, WP_016835402.1	402	6e–141	68.68
	H. seropedicae, WP_225202855.1	390	2e–136	68.06

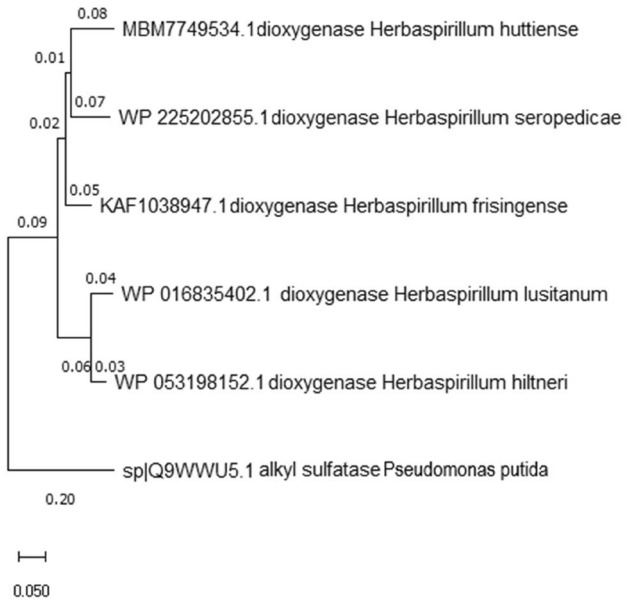

Fig. 2 The evolutionary history was inferred using the Neighbor-Joining method (Saitou and Nei 1987). The optimal tree with the sum of branch length = 0.65119288 is shown. The tree is drawn to scale, with branch lengths (next to the branches) in the same units as those of the evolutionary distances used to infer the phylogenetic tree

(Fig. 3). The culture showed a high enzymatic activity in material washed off the cell surface and in cell-free and intracellular extracts. Intracellular and surface alkylsulfatase activities were approximately at the same level (50 units/L), and extracellular alkylsulfatase activity was approximately 2.5 times higher (120 units/L) (Fig. 3).

The use of microorganisms in biosensor systems presupposes preliminary study of bacterial activity toward the substrate being tested. Therefore, we analyzed the ability of *H. lusitanum* P6–12 to utilize SDS as the only carbon and energy source. Bacteria were grown with 0–600 mg/L of SDS (Fig. 4). This range was chosen because as a rule, industrial wastewater contains anionic surfactants in the concentration range 1–10 mg/L (Fendinger et al. 1994).

The Fig. 4 data show that the growth of *H. lusitanum* P6–12 with 50 mg/L of SDS was intense and was comparable to growth without SDS. Increasing the SDS concentration to 100 mg/L decreased the number of bacteria twofold. Between 200 and 500 mg/L of SDS, growth was largely inhibited but the cells retained their viability and motility (Fig. 4). Increasing the SDS concentration to 600 mg/L resulted in cell aggregation and sedimentation.

The growth of strain P6–12 was accompanied by a decrease in the content of SDS (Fig. 4). At 50, 100, and 200 mg/L of SDS in the medium, 92.2, 77.3, and 43.8% of SDS was degraded, respectively. When the SDS concentration was increased to 400 and 600 mg/L, the degradation efficacy decreased to 21.9 and 3.7%, respectively.

Analysis with an electric sensor

Because *H. lusitanum* P6–12 has alkylsulfatase activity and can utilize SDS as a single source of carbon, we investigated whether it can be used as the sensing element of an electric sensor in SDS analysis. For this purpose, we used a sensor to examine the effect of different SDS concentrations on the electric polarizability of the bacterial cells. Figure 5a shows that under the influence of SDS, the electric polarizability of strain P6–12 changed across the range of frequencies used, as compared to the control (no SDS; Fig. 5a, line 1). For convenience of presentation, Fig. 6b shows the results for the 100, 500, and 1000 kHz frequencies. It can be seen that the minimum detectable concentration of SDS was 0.08 mg/L (Fig. 5a, line 2). Additionally, Table 2 shows the SDS-induced deviations of the analytical signal, as compared to the control (no SDS).

When SDS was used at greater than 200 mg/L concentrations, the sensor signal changed within 2–3% (Fig. 5). Therefore, we examined the sensor signal at the 0.01–10 mg/L concentration range of SDS (Fig. 6). For convenience of presentation, we give the discrete polarizability tensor values measured at the constant frequencies of 100, 500, and 1000 kHz, respectively. From 0.01 mg/L onwards, there were statistically significant changes in the signal magnitude (9, 7, and 14%), as compared to the control (Fig. 6). The set of discrete polarizability tensor values obtained approximated satisfactorily by two intersecting

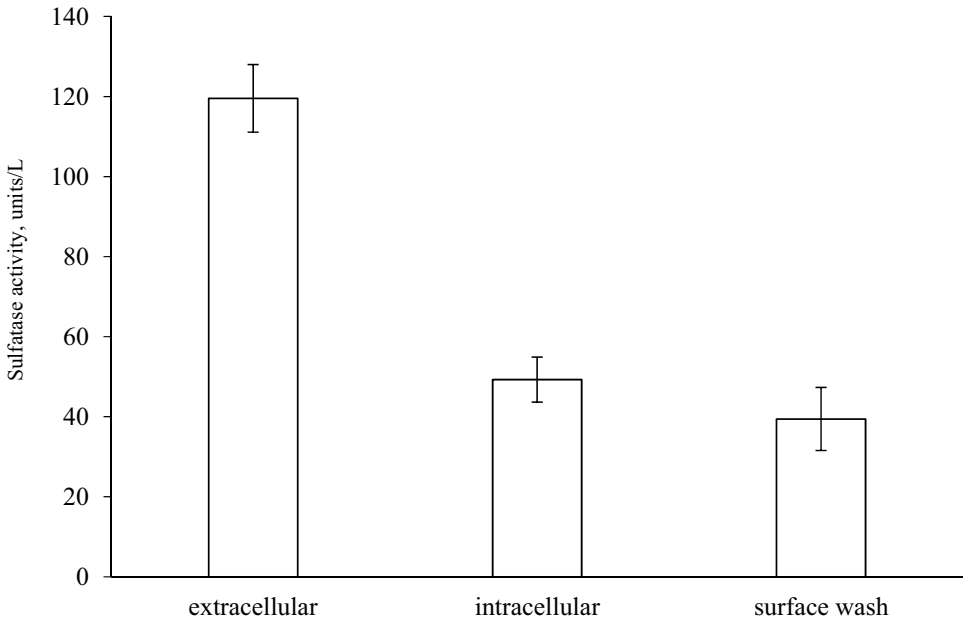

Fig. 3 Alkylsulfatase activity of *H. lusitanum* P6–12 grown in the liquid medium

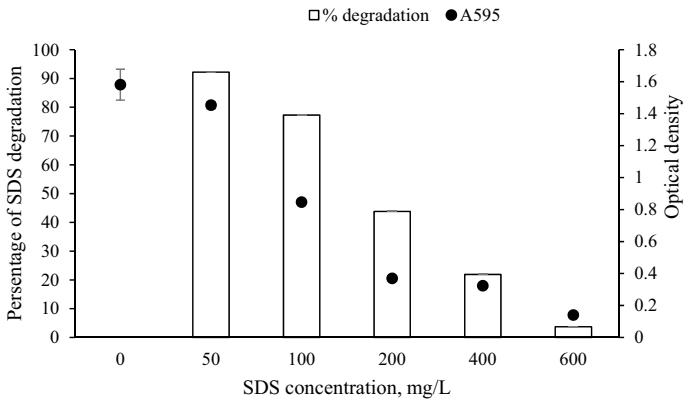

Fig. 4 *H. lusitanum* P6–12 growth in the liquid medium with different SDS concentrations (30 ^{0}C, 200 rpm, 24 h) and SDS biodegradation in the liquid medium under microaerobic conditions (pH 7.4, 30 ^{0}C, 150 rpm, 24 h)

linear functions at the concentration ranges of 0–0.1 and 0.1–2 mg/L, with coefficients of determination (R^2) values of 0.9738 and 0.9819 (data not shown), respectively. The resultant concentration dependences may be used as calibration graphs for the determination of SDS in aqueous solutions.

The interaction of SDS with the bacterial surface structures are recorded by the sensor as changes in the particle polarizability tensor. Thus, *H. lusitanum* P6–12 can be recommended as a sensing element for the determination of 0.01–400 mg/L of SDS. The determination limit of 0.01 mg/L is much lower than the official requirements for the content of SDS in potable and process water (0.5 and 1.0 mg/L, respectively). Approximately the same level of alkylsulfatases activity with a significant decrease in biomass production at SDS concentrations of 100 and 200 mg/L may be due to the action of a stressor that activates the bacterial enzymatic system involved in SDS degradation.

It follows from the Table 3 data that our whole-cell electric sensor for SDS determination has a good minimum detection limit (0.01 mg/L). In this work, the presence of SDS in the medium was recorded within an average range of 1–5 min.

Discussion

Biosensors are analytical systems that consist of a sensitive biological element and a detection system recording the concentration or activity of the analytes present in the sample. In biosensor development, it is crucial that one choose a bioselective agent (recognition element) that will ensure specific binding to the target analyte; this binding will be detected by the sensor. Microbial cells able to utilize the substrate being detected can be used as the sensing element. The preferred bioelement is a microorganism that has an

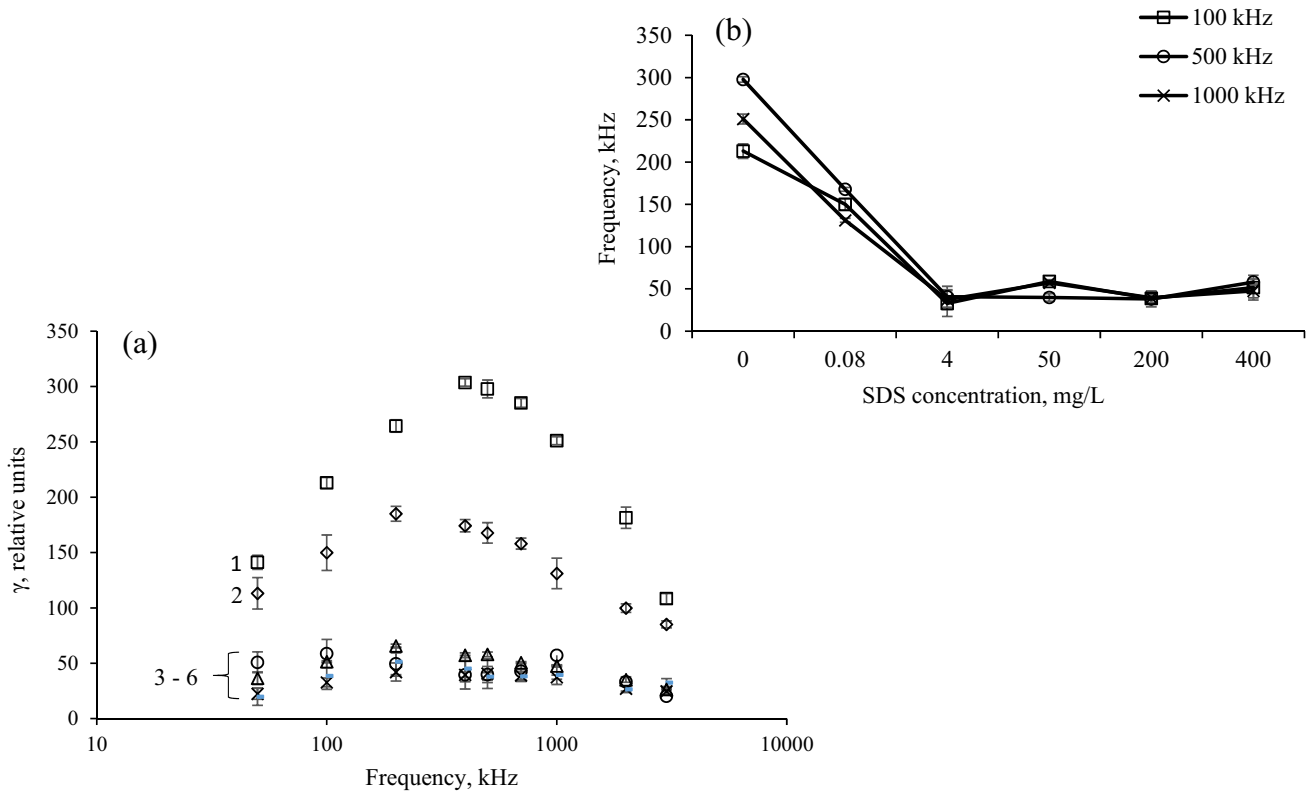

Fig. 5 Dependence of the electric polarizability of a suspension of SDS-grown *H. lusitanum* P6–12 cells (SDS concentration, mg/L: line 1, 0; line 2, 0.08; line 3, 4; line 4, 50; line 5, 200; line 6, 400) in the range of frequencies used

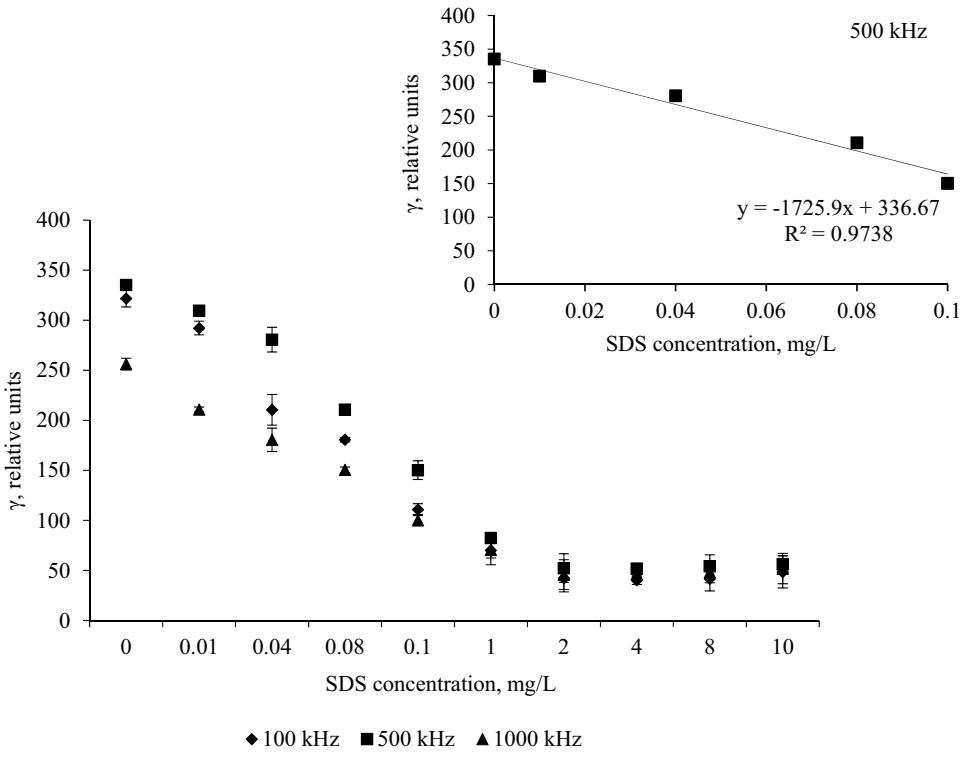

Fig. 6 Dependences of the electric polarizability of a suspension of SDS-grown *H. lusitanum* P6–12 cells (SDS concentration, 0.01–10 mg/L) at 100, 500, and 1000 kHz

Table 2 Changes in the electric polarizability of SDS-grown cells, as compared to the control (relaxation time, 4.5 s)

SDS (mg/L)	Deviation of values at different frequencies (kHz) from the control (%)								
	50	100	200	400	500	700	1000	2000	3000
0.08	19.78	29.61	29.99	42.60	43.64	44.55	47.75	44.97	21.40
4	84.11	84.62	84.07	86.96	86.32	86.33	85.10	85.08	77.12
200	85.94	81.86	80.50	85.10	87.21	86.56	84.23	85.36	69.93
400	74.10	75.82	75.17	81.14	80.50	82.26	81.04	80.57	75.64

enzymatic oxidation system for the efficient degradation of pollutants and low tolerance for their limiting effects. The use of natural (unadapted) microbial strains for these purposes will make it possible not only to ascertain pollutant effects but also to show the prospects of using the chosen strains in bioremediation.

Herbaspirillum bacteria have a plastic genome and an oxidative enzyme system, and therefore they can degrade environmental pollutants. This fact may form the basis for the design of new bacteria-based whole-cell biosensors (Tecon and van der Meer 2008; Brice et al. 2011; Hong et al. 2011.). Bacterial enzymes are under "evolutionarily optimized conditions," and in some cases, this results in highly stable analytical signals. Several studies have shown the involvement of microbial alkylsulfatases in the degradation of SDS. Alkylsulfatase (EC 1.14.11. 77; alkylsulfatase, α-ketoglutarate-dependent sulfate ester dioxygenase) is a heme-containing dioxygenase that catalyzes the cleavage of complex alkyl sulfates with medium chain lengths, such as butyl, pentyl, hexyl, heptyl, octyl, nonyl, decyl, and sodium dodecyl (SDS) sulfates (Müller et al. 2004). High degradative potential against alkyl sulfates, in particular SDS, has been shown in members of the genus *Pseudomonas*; however, the higher is the oxidative ability of a bacterial strain, the lower is the analysis selectivity when this strain is used as the sensing component (Beyersdorf-Radeck et al. 1998).

Bacterial cell surface polysaccharides, which have a large number of hydrophilic and (often) other functional groups (e.g., –COOH, $–NH_2$), are implicated in the formation of hydrate shells around cells and of the cell surface charge. In turn, the shells and the charge ensure the aggregative stability of the bacterial suspension in response to a stressor. Details of the mechanism underlying the interaction of SDS with the bacterial surface structures have not yet been clarified, but it is known that a large contribution to this process is made by hydrophobic interactions (Bhuyan 2010). Lipopolysaccharide (LPS), the major component of the outer membrane surface of gram-negative bacteria, can bind SDS molecules owing to its hydrophobicity and owing to ionic interactions with the charged groups in the core oligosaccharide. One can assume that the interaction with the core oligosaccharide would be more pronounced in R-form LPS owing to the absence of O polysaccharide chains. Such interactions of SDS and its enzymatic decomposition products with the R-LPS of *H. lusitanum* P6–12 may result in the appearance of induced charges on the bacterial surface.

Biosensors, especially those used for environmental purposes, should offer easy and quick detection of compounds (Belkin 2003; Tecon and van der Meer 2008). In this context, the use of fluorescent, electrochemical, and whole-cell sensors to determine SDS has great potential. The main criteria involved in the development of sensor systems for the analysis of toxic compounds include the specific response of bacteria, the presence of a range of linear dependence of bacterial activity toward a given substrate, the ease of preparation of active biomass, and the analysis time. Our biosensor system obviates the need for purified enzyme preparations, giving great advantages for in situ microbial ecology and largely reducing the analysis cost. Our sensor differs in principle from other whole-cell bacteria-based sensors used for SDS analysis (Table 3), because the mechanism of its action is supposedly based on the binding of SDS and its enzymatic decomposition products to the bacterial surface, which brings about changes in the polarizability tensor. An added advantage is the absence of need for bacterial cell immobilization, which largely reduces the preparation time for the sensor's sensing element.

Thus, the use of *H. lusitanum* P6–12 shows promise for use in the sensor determination of low SDS concentrations. Analysis of changes in the electrical polarizability of cells grown with SDS can both show the fact of SDS effect on microorganisms but also measure this effect by the change in the magnitude of the recorded signal.

The COVID-19 pandemic triggered a sudden increase in demand for hand and room disinfectants in hospitals, households, and industrial enterprises. Research on the lauryl sulfate market has shown a substantial increase in the production of SDS and SDS-based household cleaning products. By 2027, the market for dry sodium lauryl sulfate may be worth $620 million through its use in detergents and in personal care and beauty products. The growing urbanization, the sustained growth in the cosmetics industry, and modernization are the major factors responsible for the changing consumer preferences, which are expected to drive the global demand for sodium lauryl sulfate. It is expected that the growth of the sector of

Table 3 Summary of the methods used to detect SDS in aqueous solutions

Method	Response time (min)	Linear range of detection	References
Routine methods			
Chromatography	n.a	0.4–20 mg/L	(Pan and Pietrzyk 1995; Nair and Saari Nordhaus 1998; Bruzzoniti et al. 2008)
Potentiometry	1–3	0.3–0.63 mg/L	(Stepanets et al. 2001; Kulapina and Ovchinskii 2000)
Spectrophotometry	n.a	0,02–5.00 mg/L	(Moskvin et al. 1996a, b; Moskvin et al. 1996a; Rodenas et al. 2005; Bidari et al. 2010)
Fluorimetry	n.a	0.1–5 mg/L	(Sanchez et al. 2005; Franek et al. 2001; Ruiz et al. 2004; Villar et al. 2007)
Fluorescence sensor systems			
Cationic fluorescent probe	5–30	5–50 µM	(Chen et al. 2021)
Aggregation-induced emission-based fluorescent probe	0.66	13.07 µg/L	(Gao et al. 2017)
Recovered fluorescence signal of eosin Y and polyethyleneimine complex	30	0.4–6 µg/mL	(Wen et al. 2013)
Glutathione-stabilized gold nanoclusters and poly(diallyldimethylammonium)chloride—enhanced fluorescent system	n.a	0.2–12 µg/mL	(Zheng et al. 2014)
Polymer nanoparticles prepared with polyethyleneimine and ascorbic acid	60	0.144–2.016 µg/mL	(Fu et al. 2017)
Electrochemical sensors			
Anion exchange – based amperometry with an eosin Y/polyethyleneimine electrode	30	1–40 µg/mL	(Hao et al. 2014)
Potentiometric sensor for SDS based on an electrosynthesized polyaniline membrane	0.33	3 M	(Binas and Sevilla 2005)
Potentiometric measurements with an ion-selective electrode	0.70	1.0×10^{-5}–$1.0\times10.^{-3}$ mol/L	(Wang et al. 2011)
Electropolymerized molecularly imprinted polymer	30	0.1–1000 pg/mL	(Motia et al. 2018)
Optical sensors			
Optical sensor with a sensing membrane	10	1–50 mM	(Masadome and Akatsu 2008)
Fiber optic evanescent wave sensor	10	4–15 mg/L	(Okazaki et al. 2015)
Whole-cell bacteria sensors			
Fluorescence intensity of the *E. coli* green fluorescent protein toxicity biosensor	1	1.7 mg/L	(Ooi et al. 2015)
Transcription factor based whole cell biosensor	n.a	0.1 ppm	(Dey et al. 2020)
Amperometric sensor with *Pseudomonas rathonis* immobilized in gel agar	1.7–2.5	0.25–0.75 mg/L	(Reshetilov et al. 1997)
Sensor based on *Comamonas testosteroni* TI immobilized in gel agar	12–15	0.25–0.5 mg/L	(Taranova et al. 2004)
Whole-cell electrical sensors without immobilisation of bacteria	1–5	0.01–0.1 mg/L	This paper

na not available

fast-moving consumer goods (FMCG) will accelerate the growth of the global sodium lauryl sulfate market during the forecast period. To prevent the global environmental disaster associated with the use and deposition of SDS, researchers should develop methods for the detection of this compound, primarily in water resources.

A necessary component of current environmental research is the rapid assessment of pollution. We have described an *H. lusitanum* P6–12—based sensor system for the detection and analysis of residual SDS amounts in aqueous solutions. The system enables reliable and rapid determination of alkyl sulfates, with the minimum determination limit being 0.01 mg/L. *H. lusitanum* P6–12 offers promise for use in the determination of anionic surfactants in aqueous solutions.

A modern achievement in biological cleanup is the method of local microbial cleanup. It is based on the use of biocatalysts-active microbial degraders used either in culture or as immobilized on inert media. Bacteria unadapted to xenobiotics but showing alkylsulfatase activity, which is implicated in SDS degradation, can be used as remediation agents or as components of biofertilizers able to improve crop yields in polluted settings. *Herbaspirillum lusitanum* P6–12, able to grow with SDS as the sole carbon source, can be recommended not only as a sensing element for SDS analysis but also as a remediation agent for polluted soils. Expanding the knowledge on the microbial degradation of xenobiotics will allow the design of new technological approaches and the improvement and optimization of the existing bioremediation technologies.

Thus, gram-negative soil bacteria of the genus *Herbaspirillum* have potential as SDS degraders and can also act as bioindicators. The described sensor permits rapid determination of low SDS concentrations and obviates the need for sample pretreatment. The relatively high optimum temperature for growth on SDS is suitable for use of *Herbaspirillum* in the remediation of SDS-polluted soils.

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Author contributions NV, OG, MK, MK conceived and designed research. NV, MK and MK conducted experiments. NV, OG, MK, MK and YF analyzed data. NV, OG, MK, YF wrote the manuscript. All authors read and approved the manuscript.

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Data availability All data generated or analysed during this study are included in this published article.

Declarations

Conflict of interest The authors declare that they have no conflicts of interest.

Ethical approval This article does not contain studies conducted on human participants or animals by any of the authors.

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