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Aldehyde dehydrogenase activity promotes survival of human muscle precursor cells

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Abstract

Aldehyde dehydrogenases (ALDH) are a family of enzymes that efficiently detoxify aldehydic products generated by reactive oxygen species and might therefore participate in cell survival. Because ALDH activity has been used to identify normal and malignant cells with stem cell properties, we asked whether human myogenic precursor cells (myoblasts) could be identified and isolated based on their levels of ALDH activity. Human muscle explant-derived cells were incubated with ALDEFLUOR, a fluorescent substrate for ALDH, and we determined by flow cytometry the level of enzyme activity. We found that ALDH activity positively correlated with the myoblast-CD56+ fraction in those cells, but, we also observed heterogeneity of ALDH activity levels within CD56-purified myoblasts. Using lentiviral mediated expression of shRNA we demonstrated that ALDH activity was associated with expression of Aldh1a1 protein. Surprisingly, ALDH activity and Aldh1a1 expression levels were very low in mouse, rat, rabbit and non-human primate myoblasts. Using different approaches, from pharmacological inhibition of ALDH activity by diethylaminobenzaldehyde, an inhibitor of class I ALDH, to cell fractionation by flow cytometry using the ALDEFLUOR assay, we characterized human myoblasts expressing low or high levels of ALDH. We correlated high ALDH activity ex vivo to resistance to hydrogen peroxide (H2O2)-induced cytotoxic effect and in vivo to improved cell viability when human myoblasts were transplanted into host muscle of immune deficient scid mice. Therefore detection of ALDH activity, as a purification strategy, could allow non-toxic and efficient isolation of a fraction of human myoblasts resistant to cytotoxic damage.

Keywords: ALDH • satellite cells • myoblast • adult stem cells • skeletal muscle

Introduction

Skeletal muscle has the ability to undergo rapid repair after acute damage. This process is driven by a group of highly specialized cells – known as ‘satellite cells’ – that are found beneath the basal lamina surrounding each myofibre. While normally quiescent, satellite cells become activated upon muscle damage and start to proliferate. Then, their descendants, the myoblasts, differentiate and fuse to regenerate the damaged muscle. Beside these criteria, satellite cells can be identified by the expression of specific combinations of molecular markers, the most widely used in mouse are probably Pax7, M-cadherin and CD34 [1]. Because myoblasts can be transplanted and fuse with endogenous muscle fibres to form hybrid myotubes [2], myoblast transplantation represents a potential approach for the treatment of inherited myopathies and diseases characterized by fibre necrosis and muscle weakness [3–6]. Although other limitations, such as immune rejection or limited spread into the host tissue are also important, failure of myoblast transfer in the initial clinical trials was at least partly related to poor survival rate of transplanted myoblasts [3, 5–7]. Several approaches have been developed to reduce early loss of injected myoblasts: immunosuppressive treatment to reduce inflammation [8, 9], heat shock treatment of grafted myoblasts to
improve their resistance to apoptosis [10], control of growth factor levels to increase myoblast proliferation [11, 12] and neo-angiogenesis to reduce hypoxia and cell death [13]. One additional approach would be to select and purify a pool of myoblasts characterized by an improved survival response. Recent studies reached high efficiency of mouse myoblast transplantation after using cell-surface markers to purify a myoblast subpopulation [14]. Thus, manipulating myoblast subpopulations may have important implications for development of new treatments. However, to date, only a small number of surface markers are available to isolate human myoblasts, the most widely used being CD56 suppress (N-CAM) [15, 16]. Thus, it is important to characterize other markers of human muscle progenitors in order to identify and manipulate them efficiently.

Aldehyde dehydrogenases (ALDH) are a family of enzymes that efficiently oxidize and detoxify aldehydic products of lipid peroxidation such as 4-hydroxynonenal, which are initially generated by reactive oxygen species [17, 18]. In addition to its protective role against toxic molecules, a subfamily of ALDH is involved in retinoic acid synthesis. These retinaldehyde dehydrogenases belong to the ALDH 1, 2 and 3 subfamilies [17]. Retinoic acid binds and activates nuclear retinoic acid receptor/retinoic X receptor heterodimers to regulate the transcription of target genes important for stem cell differentiation [19]. Compelling evidence suggests that ALDH activity is associated with the stem cell status of cells in haematopoietic and nerve tissues. Indeed, selection based on high ALDH activity allows the isolation of long-term reconstituting human haematopoietic stem cells [20, 21]. This observation has been extended to other transplantable cells. Cori et al. showed that high activity of ALDH characterizes a primitive brain-derived neural stem cell population [22]. Recently, ALDH has been identified as a marker of normal and malignant human mammary stem cells [23, 24]. Therefore, it appears that ALDH confers a specific advantage to stem cells, but the molecular nature of this advantage is not clear. In summary, ALDH could enhance cell survival, stem cell renewal, proliferation or differentiation.

Here, we assessed human myogenic precursor cells (myoblasts) for ALDH functional activity. We used fluorescence-activated cell sorting (FACS) coupled to ALDEFLUOR – a fluorescent substrate for ALDH – and ex vivo and in vivo functional assays to characterize differences between human myoblast populations expressing low or high levels of ALDH. Our findings suggest that high ALDH activity is restricted to a fraction of human myoblasts and is associated with improved cell viability.

Materials and methods

Primary human muscle cell culture

Quadriceps muscle biopsies of nine human adults (mean age 34 years ± 3.02) were obtained from the ‘AFM-BTR Banque de tissus pour la recherche’ (Hôpital de la PitiéSalpêtrière, Paris, France) and from patients followed at the Service de Physiologie Clinique, Centre Hospitalier Universitaire Lapeyronie (Montpellier, France). Informed and written consent was obtained from all persons after explanation of the protocol. Skeletal muscle explant-derived cells were initially prepared. Muscle biopsies (50 mg) were scissor minced and tissue fragments were plated in collagen-coated dishes. Explants were anchored to the dish by a thin layer of Matrigel® (BD Biosciences, Le Pont De Clai, France) and maintained in growth medium composed of DMEM (Sigma-Aldrich, Saint Quentin Fallavier, France), 20% foetal calf serum (FCS) (Thermo Fisher Scientific, Illkirch, France) and 0.5% Ultroser G (Pall, France) at 37°C in 95% humidified air, with 5% CO2. After 6 to 8 days, cells migrated out of the explants. Migrating cells were enzymatically harvested using dispase (BD Biosciences) and subcultured in growth medium. On average, 30% of the total cells expressed desmin, a marker of myogenic cells (unpublished observations). To purify myoblasts, an immunomagnetic sorting system with magnetic activated cell sorter (MACS) microbeads (Miltenyi Biotec, Paris, France) directly linked to an antibody against N-CAM (CD56) was used [15]. CD56+ cells were isolated and enriched according to the manufacturer’s instructions and were at least 99% positive for desmin expression. When CD56-purified human myoblasts reached confluency, differentiation was induced by serum depletion (2% FCS). Cells were kept in differentiation medium for 2–4 days. Note that in the manuscript, ‘human myoblasts’ (hm) is commonly used instead of ‘CD56-purified’ human myoblasts. There is one exception reported in Table 1 where human myoblasts were prepared by ‘pronase digestion’ according to the protocol set up for mice, rat and rabbit satellite cells.

To determine whether ALDH activity reflects Aldh1a1 expression in human myoblasts, we used lentiviral mediated expression of Aldh1a1 shRNA (MISSION shRNA Control Transduction Particles, Sigma-Aldrich). Human myoblasts were infected on day 0 as indicated by the manufacturer. On day 2, we added 0.5 μg of puromycin in culture medium for selection of transduced cells. On day 6, fresh medium was added. Then, Western blot and flow cytometry analysis were performed. shRNA lentiviral constructs TRCN0000026498 (shAldh1a1–98) and TRCN0000026482 (shAldh1a1–82), the most efficient in inhibiting Aldh1a1, target 5’ sequences of Aldh1a1 mRNA.

Primary mouse, rat, rabbit muscle cell cultures

Briefly, satellite cells were isolated from the whole mice muscles of the paw and from rat leg muscles after enzymatic digestion by pronase [25, 26]. Rabbit satellite cells were isolated from the semi-membranous accessorius muscle as previously described [26]. Cells were plated at a density of 2 × 10^5 cells/cm² on Matrigel®-coated Petri dishes (BD Biosciences), in 80% Ham’s F10 supplemented with 20% horse serum (Invitrogen, Cergy Pontoise, France). They were maintained at 37°C in a water-saturated atmosphere containing 5% CO2 in air. After 2 days, cells were washed with Ham’s F10 and placed in complete medium supplemented with 5 ng/ml basic fibroblast growth factor. Cells were harvested at day 3.

Primary non-human primate muscle cell cultures

Cells were isolated from the appendicular muscles of 5-years old non-human primates, housed and cared for at the Boisbonne Center for Gene Therapy of the National Veterinary School of Nantes (France). Non-human primates were captive-bred Macacus cynomolgus macaques purchased from BioPrim (Baziège, France). The Institutional Animal Care and Use
Isolation of epithelial and stromal corneal cells for flow cytometry

This protocol was adapted from the dissociation procedure that uses collagenase digestion [29]. C57Bl6 mice were killed: eye balls and corneas were removed in PBS on ice. Corneal endothelium was dissected under a binocular microscope. Corneal epithelium and stroma were digested with fresh collagenase B (1 mg/ml, Roche Applied Science, Meylan, France) at 37°C with shaking for 45 min. After low speed centrifugation (250 × g, 5 min.) pellets were washed and passed through a cell strainer. Cells were analysed with a FACs Calibur flow cytometer (BD Biosciences). Data analysis was performed with CellQuestPro (BD Biosciences).

Western blot analysis

Cells were lysed in hypotonic buffer (50 mM Tris pH 7.5) and protein concentrations were determined using the BioRad Protein Assay. A total of 20 μg of total proteins diluted in 1× Laemmli buffer (100 mM Tris-Cl, pH 6.8, 4% SDS) were separated by SDS-PAGE gel electrophoresis and transferred to PVDF membranes (Millipore, Molsheim, France), blocked 1 hr at room temperature with 5% milk in PBS and probed at 4°C overnight with the following primary antibodies: polyclonal anti-Alpha-1 and anti-Alpha-1 (Santa Cruz Biotechnology, Inc., Heidelberg, Germany) diluted 1/500; monoclonal anti-tubulin (Sigma-Aldrich) diluted 1/10,000; monoclonal anti-Pax7 (Santa Cruz Biotechnology, Inc.) diluted 1/500; monoclonal anti-MHC slow and fast (Sigma-Aldrich) diluted 1/500; monoclonal anti-α-smooth muscle actin (Developmental Studies Hybridoma Bank, Iowa City, IA, USA) diluted 1/2; polyclonal anti-Myf5 (Santa Cruz Biotechnology, Inc.) diluted 1/500; monoclonal anti-destin (Sigma-Aldrich) diluted 1/1000; monoclonal anti-actinin T (Sigma-Aldrich) diluted 1/1000; monoclonal anti-caveolin-3 (BD Biosciences) diluted 1/500. Membranes were washed in PBS and incubated with a horseradish peroxidase-conjugated anti-mouse or anti-goat antibody (GE Healthcare, Orsay, France).

Immunofluorescence

Human myoblasts were fixed in PBS containing 4% paraformaldehyde (Electron Microscopy Sciences, Aguesvives, France) and permeabilized with PBS 0.5% Triton X-100. Monoclonal anti-desmin or monoclonal anti-troponin T (Sigma-Aldrich) and rabbit polyclonal anti-myogenin (Santa Cruz Biotechnology, Inc.) were added at a dilution of 1/200 before secondary incubation with Alexa 488-conjugated anti-mouse antibody (Invitrogen). For CD56 staining, living cells were successively incubated with CD56 antibodies (1/1000) and Alexa 555-conjugated anti-mouse antibody. Cells were then fixed and processed for immunofluorescence.

Flow cytometry

Cells obtained from muscle explants as ‘explant-derived’ or CD56⁺ were suspended in ALDEFLUOR assay buffer and incubated for 30 min. according to the manufacturer’s specifications (StemCell Technologies, Grenoble, France). In each experiment, an aliquot of cells was incubated under identical conditions with the specific ALDH inhibitor diethylamino-benzaldehyde (DEAB) as negative control. The amount of intracellular fluorescence was measured by flow cytometry using a FACS Calibur (BD Biosciences) and analysed using the CellQuestPro software (BD Biosciences). ALDH1A1 (top 5% of the parent population) and ALDH1A1 (bottom 5% of the parent population) were selected with FACS ARIA (BD Biosciences) (C. Duperray, Montpellier RIO Imaging facility, France).
Myoblasts were seeded onto 60 mm collagen-coated dishes, cultured in growth medium containing 10% FCS and treated with several concentrations of H$_2$O$_2$ for 24 hrs. For specific experiments, myoblasts were pre-treated (or not) twice with 50 μM DEAB, 24 hrs and ½ hr before H$_2$O$_2$ addition. Dead myoblasts were revealed by staining with ethidium homodimer-1 (2 mM, Sigma-Aldrich) and were analysed by flow cytometry using a FACS Calibur and the CellQuestPro software. Analysis of apoptosis by JC-1, caspase 3/7 and annexin labelling. JC-1 probe (Fluoprobes, Interchim, Montluçon, France) was used for the quantification of depolarized mitochondria. In non-apoptotic cells, JC-1 aggregates in the intact mitochondria resulting in red fluorescence. In apoptotic cells, JC-1 exists in monomeric form and stains cells green. Cells were collected, centrifuged and loaded with 2.5 μM JC-1 at 37°C for 15 min. Cells were washed, centrifuged and resuspended in 0.5 ml of PBS bovine serum albumin. Cell fluorescence was measured by flow cytometry. Caspases 3/7 activities were assessed by carboxyfluorescein-fluorochrome inhibitor of caspasases (FAM-FLICA) reagent (Fluoprobes, Interchim, France). Cells were collected, centrifuged and resuspended in 300 μl of 1× wash buffer and 2 μl of 150× Caspase3/7-FLICA solution were added to the cell suspension. Cells were incubated for 1 hr at 37°C. Cells were then washed, centrifuged and resuspended in 0.5 ml of wash buffer. Samples were kept on ice until FACS analysis. Phosphatidylserine externalization, an early event associated with the onset of apoptosis, was assessed with Annexin V-488 (Fluoprobes, Interchim, France). Briefly, cells were loaded with Annexin V-488 for 15 min. at 37°C. Samples were kept on ice until FACS analysis.

To determine whether ALDH activity was detectable in cells associated with skeletal muscle, we first prepared explant-derived cells from cultures of human muscle biopsies. We then incubated these cells with ALDEFLUOR, a fluorescent substrate for ALDH, and determined by FACS the level of enzyme activity. In each experiment, we incubated an aliquot of cells with ALDEFLUOR and DEAB – an inhibitor of ALDH – as a negative control. Analysis of cells derived from 5 different biopsies showed on average 20% ALDEFLUOR $^+$ cells (20.1 ± 4.1 S.E.M.) (Fig. 1A; R2 gate set using DEAB controls). Several and distinct subpopulations of precursor cells are resident in skeletal muscle, nevertheless, myoblasts, the skeletal muscle precursor cells, represent a high percentage of the cells that will migrate from the explants (data not shown; [31, 32]). To determine whether ALDH activity is associated with the myoblast fraction of cells, we plated FACS-sorted ALDH$^+$ and ALDH$^-$ 'explant-derived' cells. We looked for expression of CD56 and desmin, two markers of muscle precursor cells, by FACS analysis using anti-CD56 antibodies and by immunofluorescence with an anti-desmin antibody. Only ALDH$^+$ cells expressed desmin and CD56 in more than 99% of the cells (Fig. 1B). We further confirmed the myoblast identity of those cells by testing their capacity to differentiate into muscle fibres. We grew cells until confluence and then induced muscle differentiation by shifting to differentiating medium (2% FCS). During myogenic differentiation, myoblasts exit the cell cycle and spontaneously differentiate into a major subpopulation of quiescent multinucleated cells (myotubes) that express muscle-specific structural proteins. As expected, only ALDH$^+$ cells differentiated into myotubes as shown by troponin T expression (Fig. 1B). To validate these data, we purified myoblasts from explant-derived cells by using an immunomagnetic sorting system coupled with an antibody against CD56 [15, 32]. This technique enabled us to obtain highly purified myoblast cultures with about 99% desmin-positive cells. We then incubated myoblasts (CD56$^-$) and non-myoblast cells (CD56$^+$) with ALDEFLUOR and analysed ALDH activity by flow cytometry (Fig. 2A). We detected a population of cells with high levels of ALDH staining (not observed when cells were treated with DEAB) only in the CD56$^-$ cell fraction. Therefore, these results confirm that only myoblasts possess high ALDH activity. The percentage of ALDH$^+$ cells ranged from 38% to 64% of all myoblast cultures (51.3 ± 2.6 S.E.M.; n = 9) (Fig. 2B), indicating that about 50% of CD56-purified myoblasts exhibited low levels of ALDH activity.

To determine the functional characteristics of myoblasts with high ALDH activity compared to myoblasts with low ALDH activity, we incubated ‘CD56-purified’ human myoblasts (hm1, hm2 and hm3) with ALDEFLUOR and sorted them by FACS based on
the levels of ALDH activity. We then seeded ALDH$^{\text{high}}$ and ALDH$^{\text{low}}$ myoblasts and quantified their ALDH activity by FACS at different time-points after plating. Note that the degree of purification was not absolute because we observed 12% ± 2 of ALDH$^{\text{low}}$ myoblasts in the ALDH$^{\text{high}}$ population and 15% ± 2 of ALDH$^{\text{high}}$ myoblasts in the ALDH$^{\text{low}}$ population (Fig. 3A). The percentage of ALDH$^{\text{high}}$ myoblasts remained stable in culture. In contrast, the percentage of ALDH$^{\text{low}}$ myoblasts, which remained stable during the first 2 days in culture, progressively decrease (Fig. 3A). Such decrease of ALDH$^{\text{low}}$ cell population might be due to a selective advantage of the ALDH$^{\text{high}}$ cell population. However, the percentage of dead cells in ALDH$^{\text{low}}$ and ALDH$^{\text{high}}$ was not statistically different (data not shown) and their proliferative capacities were very similar (Fig. 3B). We then determined in FACS sorted ALDH$^{\text{high}}$ and ALDH$^{\text{low}}$ myoblasts the pattern of expression of several genes involved in myogenic cell determination such as Pax7, MyoD and Myf5 and confirmed the myogenic identity using anti-desmin antibodies. Western blot analyses showed no differences in Pax7,
MyoD, Myf5 and desmin expression between ALDH<sup>high</sup> and ALDH<sup>low</sup> myoblasts (Fig. 3C). Next we wanted to check whether sorted ALDH myoblasts could differentiate normally. We determined by Western blotting the expression of several differentiation markers: slow and fast myosin heavy chain, troponin T and caveolin-3. The level of expression for all these markers was similar in the two populations of cells (Fig. 3D). We also examined myotubes’ morphology by immunofluorescence with two differentiation markers, troponin T and myogenin. ALDH<sup>high</sup> and ALDH<sup>low</sup> myoblasts fused to form branched myotubes with aligned nuclei (Fig. 3E). Taken together these results indicate that both ALDH<sup>high</sup> and ALDH<sup>low</sup> subpopulations of myoblasts differentiated normally and similarly.

Therefore, ALDH activity is a new marker of human myoblasts and can be used to discriminate myoblasts with different levels of ALDH activity.

Aldh1a1 expression is associated with ALDH activity in human myoblasts

Two large-scale gene expression analyses of human myoblasts identified Aldh1a1 as the main ALDH protein expressed in muscle cells [33, 34]. Using Western blot analysis, we examined the expression of Aldh1a1 from CD56<sup>+</sup> and CD56<sup>−</sup> cells and from FACS sorted ALDH<sup>high</sup> and ALDH<sup>low</sup> myoblasts. Western blot revealed a strong immunoreactive 50 KD band for Aldh1a1 only in CD56<sup>+</sup> cells (Fig. 4A). Aldh1a1 was expressed mainly in FACS sorted ALDH<sup>high</sup> myoblasts and not in FACS sorted ALDH<sup>low</sup> myoblasts (Fig. 4B). Quantitative reverse transcriptase-polymerase chain reaction (RT-PCR) analysis confirmed the strongest level of Aldh1a1 expression in FACS sorted ALDH<sup>high</sup> myoblasts (Fig. 4C).

Western blot and quantitative RT-PCR analyses suggested a good correlation between mRNA level of Aldh1a1 and ALDH activity. To determine whether ALDH activity reflects Aldh1a1 expression in human myoblasts, we used lentiviral mediated expression of Aldh1a1 shRNA (Fig. 5). Three constructs targeting different regions of the gene sequence were screened for their efficacy to inhibit Aldh1a1 expression in human myoblasts. Non-target-sh lentiviral construct, that does target any human and mouse genes, was used as control. The expression of two specific shRNA, shAldh1a1–98 and Aldh1a1–82, and at a lesser extent shAldh1a1–02, resulted in a marked decrease of Aldh1a1 protein and activity (Fig. 5A and 5B), in comparison to non-infected myoblasts and to the control cells expressing non-target-shRNA. In conclusion, Aldh1a1 contributes to most if not all ALDH activity in human myoblasts.
High ALDH activity is restricted to human myoblasts

To extend our results to other species, we prepared satellite cells from skeletal muscle of 1-month and 22-month-old mice. After in vitro proliferation, we assessed the degree of myoblast purity by immunofluorescence with an anti-desmin antibody: our primary cultures were composed of about 80–85% myoblasts (Table 1). We then analysed mouse myoblasts for ALDH activity. Surprisingly, we were unable to detect significant ALDH activity in those cultures (Fig. 6A, upper panels; Table 1). Similar results were obtained using mouse myoblasts C2 cell line (Table 1).

Because the ALDEFLUOR assay kit has been developed for human cells, we asked whether it was specific for human ALDH. To test this possibility, we prepared mice primary cultures of cornea cells, a tissue rich in ALDH [17]. A significant proportion of cornea cells expressed high levels of ALDH activity (Fig. 6A, lower panels), indicating that the ALDEFLUOR assay could identify both human and mouse ALDH. We then investigated whether myoblasts isolated from adult rat, rabbit and non-human primates presented ALDH activity. Myogenicity of these primary cultures was very high as indicated by the presence of 85% to 95% desmin-positive cells. However, we did not detect ALDH activity in any of these cultures using the ALDEFLUOR assay (Table 1), suggesting that...
ALDH cells were very rare in adult mouse, rat, rabbit and non-human primates compared to human, below the level of detection by ALDEFLUOR staining.

To isolate myoblasts from mouse, rat, rabbit and non-human primates we performed a protocol of enzymatic digestion with collagenase or pronase (see Materials and Methods), whereas for human myoblasts, due to the low amount of available muscle tissue, we used the ‘explant method’ and purification with an immunomagnetic sorting system and CD56 antibodies. Such differences in protocols might select specific populations of myoblasts harbouring different levels of ALDH activity. Therefore, we have isolated human myoblasts with pronase digestion following the protocol used for mouse, rat and rabbit muscles. We obtained 85% of desmin-positive cells and found that about 30% to 40% of them were also ALDH+ (Table 1).

Hence, the method used to isolate myoblasts did not alter the percentage of ALDH+ cells. In addition, we showed that Aldh1a1 protein level was very high in mouse cornea cells and in human myoblasts whereas it was below the level of detection by Western blot in mouse, rat, rabbit and non-human primate myoblasts (Fig. 6B). Quantitative RT-PCR analysis confirmed the lowest level of Aldh1a1 expression in mouse primary myoblasts, similar to the level observed in human ALDHlow myoblasts. As expected, Aldh1a1 mRNA levels are high in mouse cornea cells and human ALDHhigh myoblasts (Fig. 6C).

These data highlight fundamental differences in levels of ALDH activity between human and mouse, rat, rabbit and non-human primate myoblasts.

**ALDH protects human myoblasts from cytotoxic effects induced by H2O2**

It has been suggested that Aldh1a1, in addition to its role in aldehyde oxidation, may contribute to the antioxidant capacity of cells [35]. To determine whether inhibition of ALDH activity affected myoblast viability, we pre-treated human myoblasts with DEAB and then exposed them to increasing concentrations of H2O2, a pro-apoptotic compound, for 24 hrs (Fig. 7). We quantified the percentage of dead cells by FACS using ethidium homodimer-1 which can only cross the damaged membranes of dead cells. Dead cells could thus be easily distinguished from healthy cells by a bright red fluorescence resulting from staining of nucleic acids by ethidium homodimer-1 [36]. Increasing concentrations of H2O2 induced accumulation of dead cells (Fig. 7A). DEAB alone did not affect cell viability. However, concomitant treatment with H2O2 and DEAB dramatically enhanced the percentage of dead cells for each concentration of H2O2. This synergistic effect occurred also in the presence of a concentration of H2O2 (i.e. 400 µM) that on its own did not affect cell viability. We observed this synergistic effect in the three different cultures of human myoblasts we tested (hm1, hm2 and hm3) (Fig. 7A). We then addressed the mechanism of cell death in human myoblasts treated with DEAB. Apoptosis is a term applied to a group of structural and molecular events that separate this type of cell death from necrosis. Depolarized mitochondria, caspases 3/7 activity and phosphatidylserine externalization, which are critical events in the apoptotic process induced by H2O2 [37], were assessed by labelling with JC-1, fluorescent caspase 3/7 substrate and Annexin V-488, respectively. A total of 400 µM H2O2 or DEAB alone did not enhance caspase 3/7 activity, the percentage of JC-1 green cells and Annexin V positive cells (Fig. 7B). However, concomitant treatment with H2O2 and DEAB dramatically enhanced the percentage of apoptotic cells characterized by caspase 3/7 activity, JC1-green staining and Annexin V positive labelling (Fig. 7B).

We then determined the resistance of ALDHhigh and ALDHlow to H2O2 treatment. We treated sorted ALDHhigh and ALDHlow from the three cultures (hm1, hm2, hm3) of human myoblasts with increasing concentrations of H2O2 for 24 hrs and determined the percentage of dead cells by FACS analysis after ethidium homodimer-1 staining. ALDHhigh myoblasts were more resistant to H2O2-induced cell death than ALDHlow myoblasts (Fig. 7C). These data suggest that ALDH protects human myoblasts against apoptotic effects induced by H2O2.
ALDH activity promotes survival of engrafted human myoblasts

We investigated whether we could correlate high ALDH activity to increased cell survival 2 days after implantation into the muscle of adult recipient. In order to track human myoblasts after transplantation, cells were labelled with intravital fluorescence marker, PKH67 dye [38, 39]. In preliminary in vivo experiments, we injected labelled myoblasts in quadriceps of scid mouse. Two days after implantation, muscle derived cells were isolated and plated for 24 hrs to recover implanted human cells and host mouse cells. Cells were then analysed by FACS and immunostaining. A distinct population of PKH67 bright was clearly identified by FACS analysis whereas no fluorescent cells were detected in muscle derived cells of control non-injected scid mice (Fig. 8A).

Immunostaining for human CD56 revealed that PKH67 labelled cells were also CD56^+/H11001 (Fig. 8B, left; CD56 in red, PKH67 in green). To confirm that PKH67 labelled cells were human myoblasts, we performed FACS analysis for CD56 and CD1001. PKH67 labelled cells were positively stained for both CD56 and CD1001 (Fig. 8B, right).

### Table 1

<table>
<thead>
<tr>
<th>Type of myoblasts</th>
<th>Myogenicity (%)</th>
<th>ALDH activity (%)</th>
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</thead>
<tbody>
<tr>
<td>Human myoblasts (CD56 purified)</td>
<td>99</td>
<td>58</td>
</tr>
<tr>
<td>Human myoblasts (pronase treatment)</td>
<td>85</td>
<td>36</td>
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<td>Mouse myoblasts (1 month)</td>
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<td>0</td>
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<td>Mouse myoblasts (22 months)</td>
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<tr>
<td>Mouse C2 cell line</td>
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<td>0</td>
</tr>
<tr>
<td>Rat myoblasts</td>
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</tr>
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<td>Rabbit myoblasts</td>
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Fig. 5 Aldh1a1 contributes to most if not all ALDH activity in human myoblasts. (A, B) Human myoblasts, transduced or not (not-inf.) with lentiviral mediated expression of control shRNA (shc) or Aldh1a1 shRNA (sh98, sh82, sh02) were analysed for Aldh1a1 expression by Western blot (A) and ALDH activity in the presence or not of DEAB by FACS (B).
Fig. 6 High ALDH activity and Aldh1a1 expression are restricted to human myoblasts. (A) Primary cultures of mouse myoblasts (upper panels) and cornea cells (lower panels) were stained with ALDEFLUOR in the presence (left panels) or not (right panels) of DEAB and analysed by FACS. (B) Protein extracts from human myoblasts (human), mouse myoblasts (mouse), mouse cornea cells (cornea (m)), rat myoblasts (rat), rabbit myoblasts (rabbit) and non-human primate myoblasts (n.h. primate) were analysed for Aldh1a1 expression. Loading control was assessed with α-tubulin (α-Tub) expression. (C) Quantitative reverse transcriptase-polymerase chain reaction analysis (RT-PCR) of relative levels of Aldh1a1 mRNA in mouse myoblasts, fractionated ALDHhigh and ALDHLow human myoblasts and in mouse cornea cells. Expression was normalized against 28s rRNA and the lowest level of Aldh1a1 expression was set at 1. Means were given ±S.E.M.; n = 3.
green). Conversely, mouse cells, identified by the absence of CD56 staining, were not labelled by PKH67. Sections of injected muscle were stained with human specific lamin A/C antibodies. Muscle of injected animals contained mononucleated human lamin A/C/H11001 cells (red in Fig. 8B, right). PKH67 labelling was restricted to lamin A/C/H11001 cells and did not diffuse to adjacent dystrophin-positive myofibres (purple in Fig. 8B). Taken together these data indicated that PKH67 did not diffuse from human to mouse cells. Therefore, PKH67 is a reliable tracking agent for identifying exogenous human myoblasts in histological sections and by flow cytometry.

We next investigated whether pre-treatment of myoblasts with DEAB could alter their survival in in vivo experiments. PKH67 labelled human myoblasts, pre-treated with DEAB, were implanted into muscle of scid mouse. Two days after implantation, muscle derived cells were harvested and analysed by FACS. As shown in Fig. 8C, pre-treatment with DEAB reduced by 40% the number of human myoblasts recovered after transplantation. We then determined the resistance to transplantation of ALDH<sup>high</sup> and ALDH<sup>low</sup> myoblasts. We incubated ‘CD56-purified’ human myoblasts with ALDEFLUOR and sorted them by FACS based on the levels of ALDH activity. Sorted ALDH<sup>high</sup> and ALDH<sup>low</sup> cells were labelled with PKH67 and implanted into quadriceps muscles of scid mouse. Muscles were harvested immediately or 2 days after implantation and the number of human myoblasts was determined by FACS analysis. The average number of ALDH<sup>high</sup> cells was moderately reduced by about 36% 2 days after implantation. In contrast, we observed a dramatic disappearance of implanted ALDH<sup>low</sup> cells, because about 75% of those cells were eliminated 2 days after implantation (Fig. 8D).

**Discussion**

Mouse and human satellite cells have been shown to be functional muscle stem cells, in that they are able to regenerate skeletal muscle and to reconstitute the satellite cell pool [40–42]. More
analysed by FACS. According to the percentage of human cells (quantified by FACS) and the total number of cells, we determined the number of human cells really present into host quadriceps muscles. 100% was arbitrary taken for control non-treated myoblasts. *** \( P \leq 0.001 \); mean values ±S.E.M. \( N = 9 \). (D) Sorted ALDH\(^{high}\) (left panel) and ALDH\(^{low}\) (right panel) human myoblasts were labelled with PKH67 and implanted into quadriceps muscles of scid mouse (\( 1 \times 10^5 \) cells per quadriceps). Muscles were harvested immediately (0) or 2 days (2) after implantation and the number of human myoblasts was determined by FACS analysis. 100% was arbitrary taken for ALDH\(^{high}\) (left panel) and ALDH\(^{low}\) (right panel) cells immediately after injection. \(* P = 0.05 \) and \(* * * P \leq 0.001 \); mean values ±S.E.M. \( N = 6 \) (point 0 days), \( N = 8 \) (point 2 days).
recently, several groups have shown that increased antioxidant capacity may represent an additional functional characteristic of stem cells [43, 44]. Expanding on this work, Urish et al. demonstrated that muscle-derived stem cells in mice were characterized by high antioxidant capacity that enhanced its regenerative capacity [45]. In this study, we identified Aldehyde dehydrogenases, a family of enzymes that efficiently detoxify aldehydic products generated by reactive oxygen species, as a marker of human myoblasts. We demonstrate that ‘CD56-purified’ human myoblasts contain a fraction of cells with different levels of ALDH activity. By inactivating Aldh1a1 in human myoblasts with lentiviral mediated expression of Aldh1a1 shRNA, we demonstrated that Aldh1a1 contributes to most if not all ALDH activity in human myoblasts. Therefore, these data clearly indicate that others ALDH isoforms do not compensate functionally for Aldh1a1. Consistent with these observations, we did not detect Aldh1a2 and Aldh3a1 expression in human myoblasts (by Western blot and QPCR, respectively; our unpublished observations). Using different approaches, from pharmacological inhibition of ALDH by DEAB to cell fractionation by FACS using the ALDEFLUOR assay, we correlate high ALDH activity to improved cell viability.

Our results established that human myoblasts exhibit ALDH activity and Aldh1a1 expression while ALDH activity and Aldh1a1 expression were below detection by ALDEFLUOR and immunostaining in mouse, rat, rabbit and non-human primate myoblasts. Similarly, in contrast to ALDH labelling in human haematopoietic tissues, detection of ALDH activity in primitive murine haematopoietic cells is still controversial [46, 47]. Analysis performed in Aldh1a1-deficient mice revealed that Aldh1a1 is dispensable for stem cell function and, in contrast to our results in human, did not contribute to ALDH activity in mouse [47]. It already has been shown that there are species-specific differences in the expression of growth factors and cell surface markers that are crucial for satellite cell function. For example, mouse myoblasts cease to proliferate in culture unless they are exposed to FGF [48], whereas human myoblasts can undergo many divisions in culture. In contrast to mouse, CD34 cells appeared at very low frequency in human myoblasts [15]. Furthermore, antibodies against CD34 cannot identify satellite cells in section of human muscle [49]. These previous results, together with our findings indicate that human myoblasts have acquired additional signalling pathways suggesting that the optimal conditions for muscle regeneration are different in each species.

A major obstacle in myoblast transplantation therapy is the poor rate of cell survival after implantation. A large percentage of myoblasts die within 48 hrs, which correlates with an acute inflammatory response at the site of injection [7]. Recent data suggested that oxidative stress which is presumably derived from damage induced by intramuscular implantation of myoblasts, may cause rapid cell death: antioxidant pre-treatment of myoblasts with CuZn-SOD improves the graft survival [50]. Conversely, inhibition of antioxidant capacity by decreasing glutathione activity reduces the regeneration capacity of muscle-derived stem cells [45]. We postulated that one of the mechanisms behind the higher viability of ALDHhigh myoblasts after transplantation may involve a resistance to oxidative stress. We showed that ALDH activity and Aldh1a1 expression were associated to resistance to H2O2 treatment, a well-established model to study muscle apoptosis associated with oxidative stress [51]. Concomitant treatment with H2O2 and DEAB, an inhibitor of class 1 ALDH, caused a synergic loss of cell viability. Because ALDH activity is mainly dependent on Aldh1a1 expression in human myoblasts, we tested whether direct inactivation of Aldh1a1 altered cell viability. We first inactivated Aldh1a1 in those cells using lentiviral mediated expression of Aldh1a1 shRNA. However, we cannot analyse our results because control cells expressing non-target-shRNA were abnormally resistant to apoptosis (data not shown). We then determined the resistance of ALDHlow and ALDHhigh myoblasts to H2O2 treatment and established that ALDHhigh myoblasts were more resistant to H2O2-induced cell death than ALDHlow myoblasts. These data suggest that ALDH activity and Aldh1a1 protect human myoblasts against cytotoxic damages.

Several groups have begun recently to address the question of muscle cell heterogeneity in mice, linking satellite cells’ phenotype to function [14, 42, 52]. Based on CD56 and desmin expression, human satellite cells and their progeny (myoblasts) were thought to be homogeneous [15]. However, increasing evidence indicates that human satellite cells are heterogeneous, based on different gene expression associated with different fates [53, 54]. For example, subpopulations of human myoblasts were characterized by their specific proliferation rate, clonogenic and differentiation capacities [53]. At least two subpopulations of human myoblasts have been reported, based on Pax7 expression [54]. Similarly, we found that ALDH- cells in muscle explant-derived cells are myoblasts but not all human myoblasts are ALDH+. However, we do not know whether ALDH is present in non-activated satellite cells because we did not manage to obtain an unambiguous endogenous Aldh1a1 signal by immunocytochemistry (unpublished observations). We observed that myoblasts with low ALDH activity generate rapidly in culture myoblasts with high ALDH activity. We checked for cell viability and found that ALDHlow did not die preferentially. As shown in Fig. 3, ALDHlow and ALDHhigh proliferate with a similar kinetic. Thus, differences in viability and proliferation cannot explain why the proportion of ALDHlow cells decreases. Our results suggest a dynamic equilibrium between these two subpopulations of cells. The molecular mechanisms and the biological significance of such phenomenon remain to be determined. However, we already know that this heterogeneity of expression is associated with a specific function because ALDHhigh cells are more resistant than ALDHlow cells in vitro, to cell death initiated by H2O2 and in vivo following cell transplantation.

Stem cell heterogeneity has been proposed also for cancer cells: only a limited number of cells harbouring specific stem cell antigens can initiate tumours when transplanted into immunodeficient mice [55]. Recently, it has been shown that cells with ALDH activity isolated from normal human breast have characteristics of mammary stem cells. Moreover, ALDH+ cells isolated from
human breast tumours include cancer stem cells [23]. Therefore, ALDH activity characterizes a subpopulation of normal and malignant mammary stem cells, supporting the concept of cancer stem cells and cellular heterogeneity.

In conclusion, our study reveals the existence of a subpopulation of human myoblasts characterized by high ALDH activity which appears to confer a survival advantage in cell grafting experiments. Because ALDH activity is observed in haematopoietic, neural, mammary and skeletal muscle stem cells, it could become a useful and non-toxic marker for the detection and isolation of a subpopulation of stem cells with interesting biological properties.

References


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